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COVER: *Peritrichus pilosulus* Scudder (Hemiptera: Lygeidae)

This rare western rhyparochromid bug was described as new to science in 1999 in the Journal of the New York Entomological Society 107:272-274. It occurs from British Columbia south to California. Only three specimens have ever been taken in BC, in *Purshia* habitat on the Osoyoos Indian Reserve near the Vincore Winery south of Vaseux Lake. This habitat was destroyed by recent fires in the Okanagan.

Illustration details:
Peritrichus pilosulus Scudder, male, dorsal habitus. Original pen and ink drawing on scratchboard by Launi Lucas, Department of Zoology, University of British Columbia.

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Antennal detection of sex pheromone by female *Pandemis limitata* (Robinson) (Lepidoptera: Tortricidae) and its impact on their calling behaviour

NAOMI C. DELURY[1,2], GARY J.R. JUDD[1] and MARK G.T. GARDINER[1]

ABSTRACT

Previous observations lead us to believe that female *Pandemis limitata* (Robinson) (0 to 24 h old) are as attractive as their pheromone gland extract to males in clean air, but are more attractive in an environment permeated with their major pheromone component (*Z*)-11-tetradecenyl acetate. Therefore, in this study, we tested the hypothesis that females can detect and/or respond to their pheromone components. Using electroantennographic detection, we found female *P. limitata* able to perceive both of their known pheromone components, (*Z*)-11-tetradecenyl acetate and (*Z*)-9-tetradecenyl acetate. Female antennal response was found to be 46.3% weaker than that of males, under identical conditions, with male antennae producing significantly higher deflections to the higher pheromone doses tested and to the plant volatile, (*E*)-2-hexanal. Observations of females in clean air versus (*Z*)-11-tetradecenyl acetate-permeated air showed no significant differences with respect to onset time, frequency or duration of calling. Females moved significantly less often in a (*Z*)-11-tetradecenyl acetate-permeated portion of a flight tunnel than in the corresponding clean-air portion.

Key Words: (*Z*)-11-tetradecenyl acetate, (*Z*)-9-tetradecenyl acetate, female electroantennography, flight tunnel, mating disruption, threelined leafroller, sprayable pheromone, microencapsulated pheromone, movement

INTRODUCTION

Although pheromone-based mating disruption of Lepidopteran species is widely employed in some agricultural systems (Thomson *et al.* 2001), questions about the behaviour of female moths in the presence of their own sex pheromone often remain unanswered. In particular, while there is evidence that some female moths perceive (Michell *et al.* 1972, Birch 1977, Palaniswamy and Seabrook 1978, Light and Birch 1979, Barnes *et al.* 1992) and even modify their behaviour (Palaniswamy and Seabrook 1978, 1985, Palaniswamy *et al.* 1979, Sanders 1987, Weissling and Knight 1996, Evenden 1998) in response to their own sex pheromone, others apparently do not (El-Sayed and Suckling 2005) and information of this type is lacking for most species where commercial use of mating disruption is under study. During our own studies of pheromone communication disruption in the threelined leafroller, *Pandemis limitata* (Robinson) (Lepidoptera: Tortricidae), it appeared that males were more responsive to "calling" females (0 to 24 h old) in a pheromone-permeated atmosphere than they were to female gland extracts, although no difference in male response was detectable between these sources in clean air (N.C.D., unpublished data). One explanation for this observed difference is that female *P. limitata* can detect their pheromone environment and adjust their behaviour in a manner that

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makes them more detectable to males than the female gland extract alone. Using electroantennograms (EAGs), we assessed the ability of female *P. limitata* to perceive their two known sex pheromone components, (*Z*)-11-tetradecenyl acetate [*Z*11-14:Ac] and (*Z*)-9-tetradecenyl acetate [*Z*9-14:Ac] (Roelofs *et al.* 1976) and asked the question, do females alter their calling behaviour in the presence of microencapsulated (MEC) *Z*11-14:Ac applied in laboratory flight-tunnel assays (Judd *et al.* 2005)?

MATERIALS AND METHODS

Laboratory cultures. Experiments were conducted with *P. limitata* from a laboratory colony maintained at the Pacific Agri-Food Research Centre, Summerland, British Columbia (BC) since 1992 that originated from wild populations collected in the Similkameen Valley of BC. *Pandemis limitata* were maintained on a modified pinto bean-based diet (Shorey and Hale 1965) at 25 °C under a 16:8 h L:D photoregime. Pupae were removed from diet, sexed and placed individually in 150 ml plastic cups provided with a wet cotton wick until adults eclosed. Male and female moths were isolated from each other in separate environmental chambers (25 °C, 65% r.h. with a 16:8 h L:D reversed photoregime).

Female and male perception of synthetic pheromone. Perception of synthetic pheromone by female and male *P. limitata* was measured using EAGs. Our EAG system consisted of an IDAC-02 computer-coupled data acquisition board, an INR-02 EAG-SSR system and AutoSpike software (Syntech, Hilversum, The Netherlands). Antennae excised from 0 to 24 h old females or males were mounted individually inside 10 µl glass capillaries containing silver-coated wire recording electrodes. Glass tubes were pulled at 300 °C on a Narishige (Model PN-30, Tokyo, Japan) micropipette puller. The distal antennal segment was removed and that end of the remaining antenna inserted into the indifferent electrode capillary. As a result, each antenna was suspended between two glass capillary electrodes filled with insect Ringer's solution (DeLury *et al.* 1999). Each antenna was challenged with six doses, in ten-fold increments from 0.1 ng to 10 µg, of *Z*11-14:Ac (97.5% purity, Sigma Chemical Company, St. Louis, MO), *Z*9-14:Ac (97% purity, Regine Gries, Simon Fraser University, BC), and a 94:6 ratio blend of both, respectively. The amount of *Z*11-14:Ac remained equal between the treatments of the main component and the blend; female antennae were also exposed to 100 µg doses from both *Z*11-14:Ac and the blend. Each antenna was exposed to each dose of each of the three compounds in order of increasing concentration. Treatments were puffed (200 ms; 10 ml per s) over each antenna, beginning with the lowest dose (0.1 ng) of each of the three compounds, which were presented in random order at each incremental dose. Baseline antennal responses were established using HPLC-grade hexane (two puffs) and a puff of the plant volatile, (*E*)-2-hexanal in paraffin oil, before and after each pheromone test concentration. All stimulus puffs were applied at 30 s intervals. All chemical stimuli were dispensed in 10 µl aliquots onto folded Whatman #5 filter paper (3 cm × 2 cm), placed in pipette tubes and connected to the puffer. The procedure was repeated using six female antennae and four male antennae.

Normalized percentage antennal deflections were calculated using the plant volatile response as the standard. Data were analyzed by one-way analysis of variance (ANOVA) followed by Dunnett's test to compare each treatment mean to the hexane control. All experimental error rates were set at $\alpha = 0.05$. Comparisons between female and male antennal responses at each dose were conducted on individual antennal deflections (mV) by first performing a two-way ANOVA, where the factors were sex and pheromone stimulus. A sex × pheromone stimulus interaction term was in-

cluded in the model. Following a significant ANOVA, linear contrasts using *t*-ratio statistics were used to make paired comparisons of mean antennal deflections for males and females at each dose. Experiment-wise error rates for these multiple-paired comparisons were fixed at 5% by adjusting α values for individual comparisons using the Bonferroni inequality. All statistical tests were performed with JMP 5.1 (2003).

Female behaviour in a pheromone-permeated environment. Female moths were placed in clean air or a pheromone-permeated environment and observed in side-by-side comparisons. To accomplish this, a sheet metal divider (73.3 cm × 37.4 cm) was installed vertically in the middle of the upwind portion of a pulling-style flight tunnel [75 cm wide × 73.3 cm high × 187 cm long flight section] covered with a replaceable 4 ml transparent polyester 'skin' (3M Canada Company, London, ON) (Judd *et al.* 2005), creating two identical isolated chambers (upwind area = 36.65 cm^2). The upwind cross-sectional surface was constructed of a set of replaceable horizontal sheet metal panes (73.3 cm × 3.7 cm with a 0.7 cm bend for stability) onto which pheromone was applied, with an untreated G200 Filtrete® with 0.5 oz Coverweb® (3M Canada Company, London, ON) that smoothed airflow, sealing the tunnel ca. 20 cm upwind of the panes. Smoke tests confirmed that air was drawn directly into each chamber at the upwind end and immediately moved downwind in a linear fashion through the tunnel. After observing females in both chambers in clean air, one chamber was randomly chosen to receive pheromone (10 mg ai · m^{-2}), which was applied to horizontal metal panes forming the upwind end of the tunnel. As a precaution, pheromone was not applied to a 1.5 cm strip immediately adjacent to the untreated chamber, creating a buffer to ensure no pheromone entered the clean-air portion of the tunnel. Untreated panes were shielded during all pheromone applications and the tunnel was allowed to ventilate for a minimum of 18 h between replicates. Pheromone treatment consisted of Phase I MEC *Z*11-14:Ac (3M Canada Company, London, ON) applied at a rate equivalent to 10 mg ai · m^{-2} to the cross-sectional area of the tunnel as described in detail by Judd *et al.* (2005).

Virgin female moths (0 to 24 h old) were chilled for 5 min at 2 °C and transferred individually into stainless steel mesh observation chambers (4.5 cm W × 3.5 cm H × 5 cm D). Chambers were stacked vertically to form a series of five individually-caged females. Treated and control portions of the tunnel each received one series of five individually-caged females ca. 2.5 h before scotophase, for a total of 15 females in each treatment. Females were observed for calling (raised wings with protruding abdomen) and movement (physically walking in the chamber) every 15 m until the initiation of scotophase, when they were observed continually for 3 h. Data gathered before scotophase was used solely for determination of onset of calling behaviour and was not included in any other analysis.

Frequencies, or the number of discrete observations, of calling and movement were analyzed separately using a two-factor ANOVA where treatment (*Z*11-14:Ac- and clean-air) and vertical position of female were the factors (JMP 5.1 2003). Onset and duration of calling were analyzed using a two-sided Wilcoxon Signed Ranks Test (JMP 5.1 2003), as the assumption of normality was not met for these data. The tunnel was assessed for positional bias between the two sides with respect to each behaviour before the application of pheromone and resulting data were analyzed as above.

RESULTS

Female and male detection of synthetic pheromone. EAGs confirmed that adult female *P. limitata* can perceive both components of their sex pheromone ($F_{21,224}$ = 19.40; $P < 0.0001$). However, antennal deflections significantly differed from those of the hexane control only for doses of 10 µg or greater for *Z*11-14:Ac and the 94:6

ratio of *Z*11-14:Ac: *Z*9-14:Ac (Fig. 1A). Response to *Z*9-14:Ac differed from the hexane control at each of the extreme doses tested, 0.1 ng and 10 µg, but did not differ from any of the doses in between (Fig. 1A). Antennae of male *P. limitata* were more sensitive to the pheromone components than female antennae (with the exception of 0.1 ng *Z*9-14:Ac), responding with deflections significantly higher than those to hexane at 1 µg for each individual compound and as low as 100 ng for the blend (Fig. 1B). Male response to the plant volatile was also significantly higher than their response to hexane (Fig. 1B). Females had significantly ($F_{1,19}$ = 125.85; $P < 0.0001$) lower antennal deflections (least squares mean ± SE: -6.13mV ± 0.30mV) when compared to males (least squares mean ± SE: -11.41mV ± 0.36mV). Comparisons of female and male antennal responses to individual doses showed that females had significantly ($F_{20,352}$ = 25.11; $P < 0.0001$) lower antennal deflections for 10 µg *Z*9-14:Ac, and for both 1 µg and 10 µg of *Z*11-14:Ac and the 94:6 ratio of *Z*11-14:Ac: *Z*9-14:Ac. Male antennal response to the plant volatile was also significantly higher than the female's ($F_{20,352}$ = 25.11; P = 0.00134).

Female behaviour in a pheromone-permeated environment. Observations of females in clean air and *Z*11-14:Ac-permeated environments showed that there were no significant differences in onset of calling ($P > 0.934$), frequency of calling ($F_{1,24}$=0.6859; $P \geq 0.4157$), or duration of calling ($P > 0.890$) (Fig. 2) among females. However, females moved significantly less in the *Z*11-14:Ac environment ($F_{1,11}$=5.0999; $P \leq 0.0452$) than in clean air (Fig. 2). There was no observed positional bias in the tunnel for either chamber with respect to any of the observed behaviours [($P \geq 0.152$), ($F_{1,14}$=3.5383; $P \geq 0.0809$), ($P \geq 0.193$) and ($F_{1,10}$=1.1703; $P \geq 0.3047$) respectively]. No vertical positional effects were observed for frequency of calling or movement [($F_{1,24}$=0.9835; $P \geq 0.4352$) and ($F_{1,11}$=0.8725; $P \geq 0.5107$) respectively].

DISCUSSION

We have found that adult female *P. limitata* (0 to 24 h old) are able to perceive both of their pheromone components; however, significant responses to the main component alone or in a blend were only detected for doses of 10 µg or greater, in contrast to males, which responded to 1 µg of the individual compounds and to 100 ng of the blend. Interestingly, female antennae gave a significant response to only the lowest (0.1 ng) and the highest (10 µg) doses of the minor component *Z*9-14:Ac. It is possible that as treatments were presented randomly in increasing concentrations, 0.1 ng *Z*9-14:Ac would have contacted a relatively 'fresh' antenna compared to subsequent doses, and as *Z*9-14:Ac is the minor pheromone component, occurring at approximately 6 to 9% in the female gland (Roelofs *et al.* 1976), the antennae may be able to perceive it at a lower level than *Z*11-14:Ac; however, a similar phenomenon was not observed for the blend or for the males.

The ability of female *P. limitata* antennae to perceive their own pheromone components, albeit at high doses, is not surprising as this has been shown for other tortricids. Palaniswamy and Seabrook (1978) noted that female *Choristoneura fumiferana* (Clemens) (Lepidoptera: Tortricidae) displayed threshold response levels corresponding to pheromone concentrations above which various behavioural patterns became evident, such as increased walking, extension of the ovipositor and antennal grooming. Ross *et al.* (1979) examined response of female *C. fumiferana* antennae to differing doses of pheromone and with increasing age, finding that females have a higher threshold for pheromone response than males and that female antennae are at their peak responsiveness in 3 to 6 d old insects. Higher response thresholds may be explained by the fact that antennae of female *C. fumiferana* have one-third to one-half the number of sensilla trichodea as

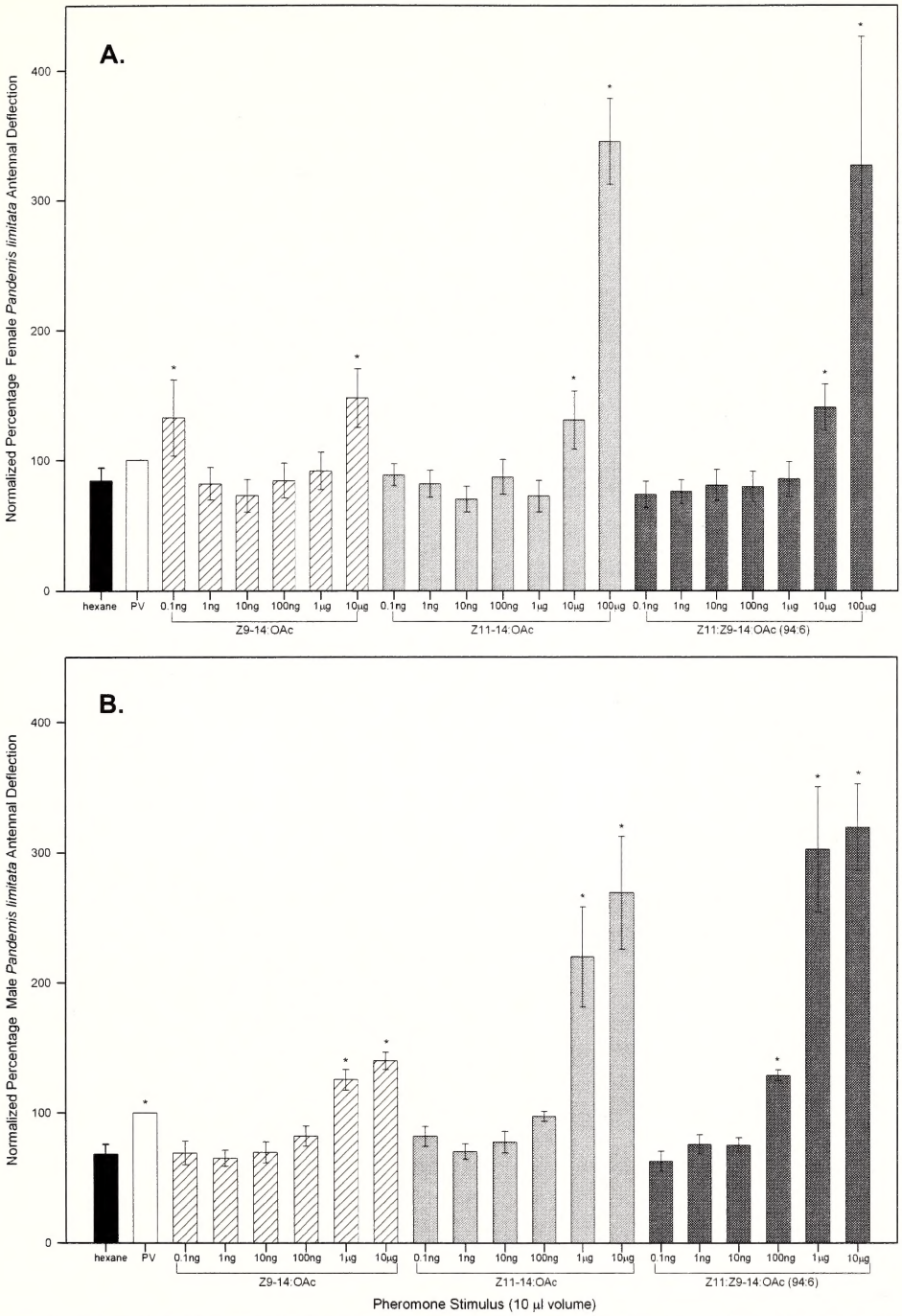

Figure 1. Mean (± SE) normalized electroantennogram responses of antennae from 0 to 24 h old A) female (n = 6) or B) male (n = 4) *Pandemis limitata* to synthetic (*Z*)-11-tetradecenyl acetate (Z11-14:OAc) or (*Z*)-9-tetradecenyl acetate (Z9-14:OAc), or both in a ratio of 96:4. (*E*)-2-hexanal was puffed over each antenna before and after each pheromone source for the purpose of normalization of each antenna over time and HPLC-grade hexane was puffed over each antenna at the beginning and end to establish a base response. All stimulus puffs were spaced by 30 s intervals. Normalized percentage deflections were calculated using the plant volatile (PV) response as the standard. Asterisks indicate a significant difference ($P \leq 0.05$) from the hexane base response as determined by an ANOVA followed by Dunnett's test, treatment versus hexane ($\alpha = 0.05$).

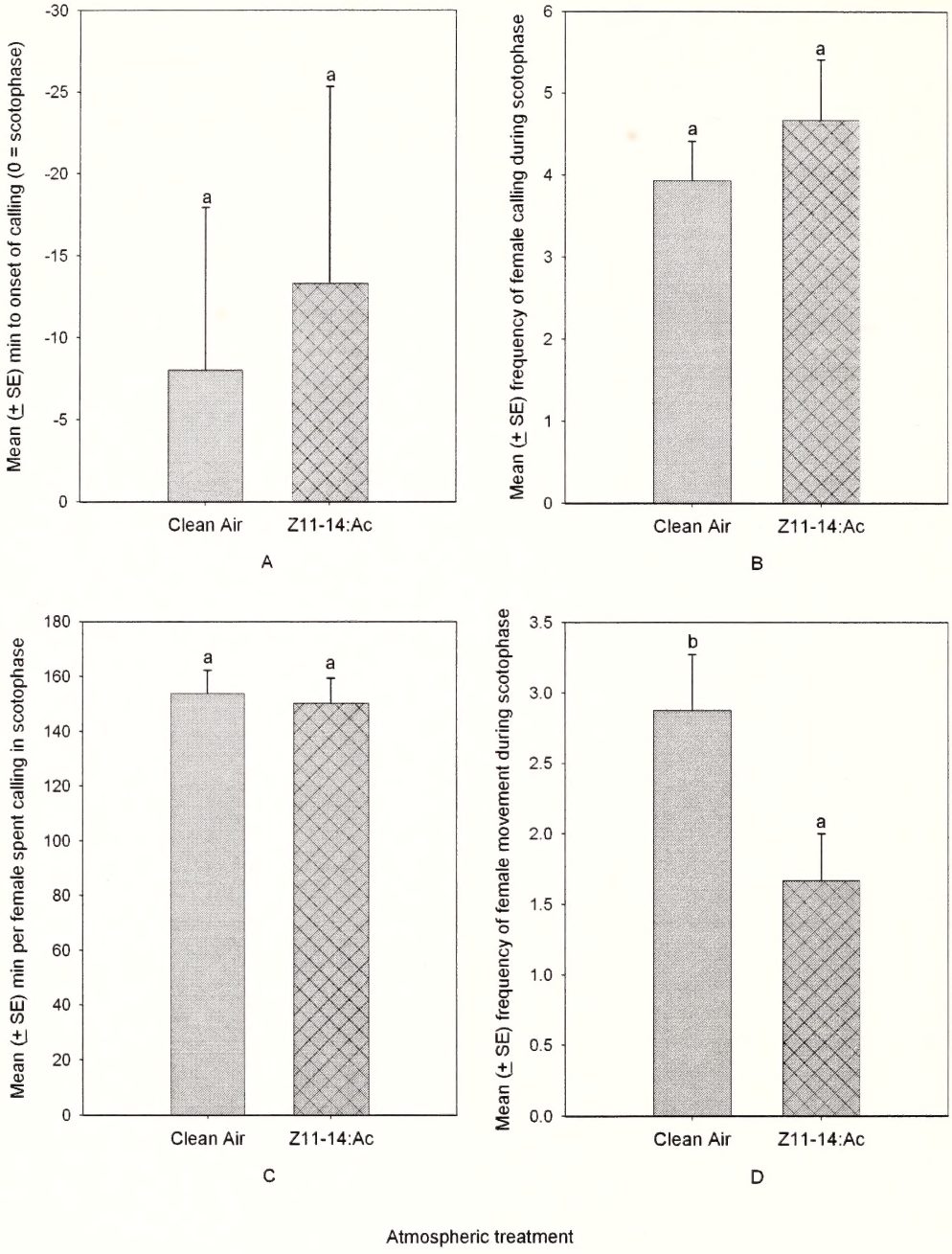

Figure 2. Mean (+ SE) duration (A and C) or frequency (B and D) of 0 to 24 h old female *P. limitata* in either side of the divided permeation tunnel, exposed to either clean air or (*Z*)-11-tetradecenyl acetate (Z11-14:Ac) released from 3M microcapsules applied to the upwind portion of one side of the tunnel. Individually-caged females were placed in the tunnel (n = 15), immediately before pheromone application ca. 2 h before scotophase. Females were observed every 15 m from application until initiation of scotophase for onset of calling, and continuously once scotophase began for frequency and duration of calling and frequency of movement. For each behaviour observed, bars with the same letter are not significantly different ($P > 0.05$).

male antennae (Albert and Seabrook 1973). In *Trichoplusia ni* Hübner (Lepidoptera: Noctuidae), Light and Birch (1979) found that antennal response in females was 25% that of males. Our results show that antennal responses in female *P. limitata* over a wide range of pheromone doses is 46.7% less than that of males, with significant differences observed at high doses for all compounds tested. A difference was even observed for the plant volatile.

Simultaneous observations of females in clean and pheromone-permeated air, from ca. 2 h before initiation of scotophase until 3 h into scotophase, showed no difference in onset of female calling time (Fig. 2A). However, a larger sample size may reveal that females in environments permeated with *Z*11-14:Ac do initiate calling earlier as there was a large variance among females. No differences were detected with respect to duration or frequency of calling once scotophase began (Fig. 2B, C). We did not have the ability to measure female output of pheromone directly in a background of *Z*11-14:Ac, therefore we must rely on observations of behaviour to give us an indication of female response. We did not observe changes in frequency or duration of calling, as one might expect if females were altering concentrations or outputs of their effluvia to compete with the level of compounds perceived in the background (N.C.D., unpublished data). Of course, these changes, as well as a change in onset of calling, may not be observable in the first 3 h of scotophase, only becoming apparent after the female has been in the environment continuously for more than 24 h (Palaniswamy and Seabrook 1985). In fact, studies on female *C. fumiferana* have found that female EAG responses to their sex pheromone increase with age at least until three days after emergence (Palaniswamy and Seabrook 1978), indicating that females may become more sensitive to their pheromone over time. As we only looked at response in 0 to 24 h old *P. limitata* females for a 5 h period, we would not have observed such a phenomenon if one existed.

Our observations showed females moved less often in air permeated with Z11-14:Ac than females in the clean air (Fig. 2D). It is possible that the tendency to remain still in the pheromone background may translate, in time, to increased calling, as observed in *C. fumiferana* (Palaniswamy and Seabrook 1985). Sanders (1987) found that although pheromone in the background clearly increased flight activity of both virgin and mated females, virgin females remained inactive for 48 h after emergence, even in the presence of the pheromone. As we observed increased movement of females in clean air, this does not appear to be happening in *P. limitata*, although observations over time would determine if the level of activity increases with age.

Similar to our results in *P. limitata*, El-Sayed and Suckling (2005) found that permanent exposure of female *Eupoecillia ambiguella* (Hübner) (Lepidoptera: Tortricidae), *Lobesia botrana* (Denis and Schiffermüller) (Lepidoptera: Tortricidae), or *Spodoptera littoralis* (Boisduval) (Lepidoptera: Noctuidae) to their main pheromone compounds did not alter the timing, duration or frequency of calling. In addition, Weissling and Knight (1996) found that the temporal pattern of calling in *Cydia pomonella* (L.) (Lepidoptera: Tortricidae) was unaffected by the major component of their pheromone; however, the likelihood of calling was increased. Nevertheless, other species have been found to initiate calling earlier in pheromone environments, such as female *C. fumiferana* (Palaniswamy and Seabrook 1985). At the other extreme, Evenden (1998) observed a delay in calling for female *Choristoneura rosaceana* (Harris) (Lepidoptera: Tortricidae) in field plots treated with their complete pheromone blend, followed by a potential reduction in time spent calling compared to their counterparts in clean air. It is important to remember that our experiments, as well as those of El-Sayed and Suckling (2005), did not use the complete pheromone blend in the background, which could have a greater impact on female response.

While experiments described here do

not encompass all of the potential factors that may impact females in a pheromone environment, such as background pheromone dose, completeness of pheromone blend, age of females or changes in mating status, our results are relevant to previous work on disruption of pheromone communication in *P. limitata* (Judd *et al.* 2005; N.C.D., unpublished data). The presence of the major pheromone component (*Z*11-14:Ac, applied at 10 mg ai · m^{-2}) does not appear to cause female *P. limitata* to change their calling behaviour, except with respect to frequency of movement, during the first 3 h of the first scotophase. As such, alternative explanations, such as mode of delivery of the gland extract, as well as the use of nonchemical cues to attract males like sound or vision (Castrovillo and Cardé 1980), or even chemical cues not associated with the sex pheromone gland, need to be explored to determine what cues become important to males as they search for female moths in pheromone-permeated environments (N.C.D., unpublished data).

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Moisture tempers impairment of adult *Otiorhynchus sulcatus* (Coleoptera: Curculionidae) climbing ability by fluoropolymer, talc dust, and lithium grease

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ABSTRACT

As part of a project to develop tools for the physical exclusion of flightless root weevils, adult black vine weevils (BVW), *Otiorhynchus sulcatus* (F.), were placed in open enclosures with smooth walls of glass, plastic or aluminum to test their ability to escape by climbing. Enclosure walls were left untreated or were treated with substances known to reduce insect climbing ability: fluoropolymer, powdered talc and lithium grease. No BVW escapes were observed under dry conditions, but all treatments allowed some escapes under wet conditions, suggesting that moisture helps BVW adults scale treated surfaces. The results help explain the ability of root weevils to overcome physical barriers under field conditions.

Key Words: black vine weevil, insect barrier, physical control, root weevil

INTRODUCTION

Like other root weevils, the black vine weevil (BVW), *Otiorhynchus sulcatus* (F.), feeds on roots as a larva, leaves as an adult and disperses by walking during the wingless adult phase. The biology and control of BVW was reviewed by Moorhouse *et al.* (1992).

Flightless root weevils could be particularly susceptible to physical control by exclusion. While hardly a new strategy (Feytaud 1918), physical control has recently been the subject of some interest (Vincent *et al.* 2003). An aluminum fence with a band of lithium grease (Cowles 1995, 1997) or fluoropolymer-coated tape (Bomford and Vernon 2005) near the upper edge can limit root weevil movement. Also effective is a portable plastic trench, designed to exclude Colorado potato beetle, *Leptinotarsa decemlineata* (Say) (Hunt and Vernon 2001). Both the fence and the trench have reduced root weevil immigration into strawberry plots by about two-thirds (Bomford and Vernon 2005). Sticky bands and fluoropolymer-coated tape on shrub stems are both recommended to reduce adult feeding on leaves (Antonelli and Campbell 2001).

Like other insects, root weevils climb using a combination of tarsal claws to hook textured surfaces and adhesive pads on their tarsomeres to attach to smooth surfaces. These adhesive pads consist of densely packed setae, each with a terminus a few μm in diameter that attaches to the surface through weak van der Waals and capillary forces (Arzt *et al.* 2003, Gao and Yao 2004). The sum of these weak forces can support the insect only if a sufficient proportion of the setae contact the surface.

Insect tarsi cannot adhere to surfaces with sufficient micro texture to prevent a large proportion of setae from making contact, but insufficient macro texture for tarsal claws to grip. Lithium grease is one such surface, consisting of an open, fibrous crystal matrix that holds tiny (~1 μm) oil droplets (Wilson 1964); fluoropolymers have similar properties (Hougham 1999). Smooth surfaces coated with fine, loose

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dust particles are similarly difficult for insects to climb because their tarsi adhere to dust particles, which slip away from the surface (Boiteau and Vernon 2001). Smooth dusted surfaces have shown potential as physical barriers to Colorado potato beetle (Boiteau *et al.* 1994, Boiteau and Osborn 1999), and root weevil (M.K.B., personal observation) movement.

This paper describes laboratory and field studies testing the influence of surface treatment and moisture on the ability of adult BVW to climb materials that could be used to construct physical barriers to root weevil migration. The results are intended to aid in the development of physical control tactics for root weevil management.

MATERIALS AND METHODS

Test insects. BVW adults were collected from an apple rootstock nursery and home garden near Vancouver, BC in late summer and early fall. Weevils were held for no more than 30 days at 20 °C under a 16:8 h L:D photoregime in clear plastic cages containing potted strawberry, *Fragaria x ananassa* (Duchesne) plants as a food source.

Glass surface treatments (dry). Eleven 250 mL glass Erlenmeyer flasks were washed and dried. One end of a length of surgical tubing was placed in each flask to allow air to escape as it was dipped upside-down in liquid fluoropolymer (Insect-A-Slip, BioQuip, Rancho Dominguez, CA) (four flasks), or powdered talc (four flasks), evenly coating the top 3 cm of the neck with the dip treatment. Excess talc and fluoropolymer were shaken off, and the fluoropolymer was allowed to dry to a hard, smooth finish. Three remaining flasks were left untreated as controls (unequal replication reflects flask availability). Flasks were randomized, five BVW adults were placed in each and all flasks were placed in an incubator held at 20 °C and 20% RH under a 16:8 h L:D photoregime. The number of weevils in each flask was recorded after 0.5 h and all escapees were removed from the incubator. The number of weevils remaining in each flask was recorded again after 24 h when the experiment was terminated. Data were analyzed by one-way ANOVA for unequal number of replicates and treatment means were separated by Tukey's honestly significant difference test (JMP Version 4.0.4, SAS Institute 2001).

Outdoor plots. Three, one m square enclosures, constructed from aluminum gutters (75 mm deep by 120 mm wide) sealed at all joints with hot glue, were sunk into freshly-tilled soil so that the soil surface was even with the upper lip of the gutter. The soil inside each enclosure was covered with a square of landscape fabric with its edges screwed to the inner gutter wall. One litre of 1:1 water:dormant oil emulsion was poured into each gutter.

Each enclosure was randomly assigned to one of three treatments: The landscape fabric pad was separated from the gutter by: 1) a 20 cm high aluminum fence with fluoropolymer-coated tape (EnviroSafe, Professional Ecological Services, Victoria, BC) attached to the upper edge of the inner surface (fence); 2) a portable plastic trench (Hunt and Vernon 2001) coated inside with dormant oil (trench); or 3) no barrier (control).

Two days after plot setup, marked BVW adults were released in the centre of each enclosure at 2200 h, a time of high activity among wild specimens observed in the area. A flashlight was used to observe weevil movement at five min intervals for one h after release. Weevils that entered the aluminum gutter and became trapped in the dormant oil emulsion (successful escapes) were recorded during the first hour and again the following morning at 1000 h. The experiment was conducted in the same plots three times (13, 18, and 20 August 1997), with ten BVW per treatment in the first replicate and 20 in the others. Hourly RH readings recorded at the Vancouver International Airport (6 km from study site) during each observation period were used to esti-

mate the ambient RH range for each replicate (Environment Canada 2005).

A two-way ANOVA was used to test for treatment and replicate effects on weevil escape rates after one and 12 h, and for interaction between factors (JMP Version 4.0.4, SAS Institute 2001). Means were separated by Tukey's HSD test.

Plastic surface treatments (wet vs. dry). Forty, 35 mL black plastic film canisters (30 mm diameter by 50 mm deep) were washed, dried, and randomly assigned to one of four treatments: ten were untreated controls; ten were dusted with powdered talc; ten were coated with liquid fluoropolymer; and ten had a 2.5 cm band of white lithium grease applied to the inner top edge.

The following day, half of the canisters from each group were rinsed with water and then emptied, leaving droplets inside. These were placed in a sealed plastic container containing an open water source to create a saturated environment. The remaining unrinsed canisters were placed in an identical container without a water source (ambient RH: 50-74%, Environment Canada 2005) and left open to allow air circulation. Canister order was randomized within each container.

Two BVW adults were placed at the bottom of each canister. The number of weevils remaining in each canister was recorded and escapees were removed at 0.5 h intervals for 3.5 h. Canisters were not treated on the outside, so re-entry was possible, but never observed. ANOVA was used to test for treatment effects within each container and means were separated by Tukey's HSD test (JMP, Version 4.0.4, SAS Institute 2001). A t-test was used to compare escape rates between containers for each treatment.

Plastic surface treatments (saturated vs. ambient RH). Eighteen, 290 mL plastic cups (50 mm diameter at base, 70 mm diameter at opening, 100 mm deep) were randomly assigned to one of three treatments: six were untreated controls; six had a 2.5 cm strip of white lithium grease applied around the inner top edge; and six were dusted with powdered talc.

Three BVW adults and a moist cotton swab were placed in the bottom of each cup. Cups from each treatment were evenly divided into two identical plastic tubs, each containing a damp cloth. One tub was sealed to create a saturated environment in which condensation formed on the plastic cups; the other tub was left open to allow air circulation and prevent condensation (regional ambient RH: 67-95%, Environment Canada 2005). Tubs were held at 20 ° C for 20 h. Any weevils that escaped from their cups were removed from the tubs at hourly intervals for the first six hours and then every other hour thereafter until the study was terminated. The mean number of escapes per cup was calculated for each treatment in the open and sealed containers. ANOVA was used to test for treatment effects within each container and means were separated by Tukey's HSD test (JMP Version 4.0.4, SAS Institute 2001). A t-test was used to compare escape rates between containers for each treatment. The time required to escape under each combination of conditions was estimated by Kaplan-Meier analysis and a Wilcoxon t-test was used to test for differences in escape times between treatments (JMP Version 4.0.4, SAS Institute 2001).

RESULTS

Glass surface treatments (dry). Almost all (93.3 ± 3.3%, $n = 3$) weevils in the control flasks escaped, but none (0.0 ± 2.9%, $n = 4$) escaped from flasks treated with talc dust or fluoropolymer, demonstrating a strong treatment effect ($F_{2,8} = 285$, $P < 0.001$). All escapes from the control flasks occurred within the first 30 min of the 24 h observation period. Weevils in the fluoropolymer treated flasks were frequently observed walking up the glass to the fluoropolymer strip and were occasionally able to climb part-way over this strip before falling. When the experiment was

terminated, approximately half of the weevils in the fluoropolymer treated flasks were on the flask walls. Weevils in the talc treated flasks showed much less ability to scale the glass walls and were all at the bottom of the flask at the end of the experiment.

Outdoor plots. Treatment and replication both affected weevil escape rates ($F_{2,141}$ = 189 and 25, respectively; $P < 0.001$) and an interaction was found between these factors ($F_{4,141}$ = 12; $P < 0.001$). Almost all weevils left control plots over the course of all replications (Figure 1), but escapes from plots surrounded by physical barriers only occurred in the third replication, conducted under light rain and high humidity conditions. Under the drier conditions of the first two replications weevils quickly climbed the aluminum fence to the lower edge of the fluoropolymer-coated tape and were unable to climb further for the duration of the test. Most weevils surrounded by plastic trenches fell into the trenches and none emerged. Under the wet conditions of the third replication the first of 20 weevils was able to walk onto the fluoropolymer within 5 min of its release. Within 20 min, four more had achieved this feat, two had reached the top of the aluminum fence and one had crossed the trench. Statistical comparison of the replications showed a higher escape rate from the fenced treatment in the third repetition after 12 h ($F_{2,47}$ = 26; $P < 0.001$), but not from the trenched treatment ($F_{2,47}$ = 2.5; P = 0.09).

Plastic surface treatments (wet vs. dry). Under dry conditions all weevils escaped from untreated canisters but none escaped from those treated with talc, fluoropolymer, or white lithium grease (Table 1). Talc lost its dusty character under wet conditions, allowing more escapes (Table 1). The dried fluoropolymer reverted to a liquid state in the presence of moisture, clumping on tarsi and allowing only one

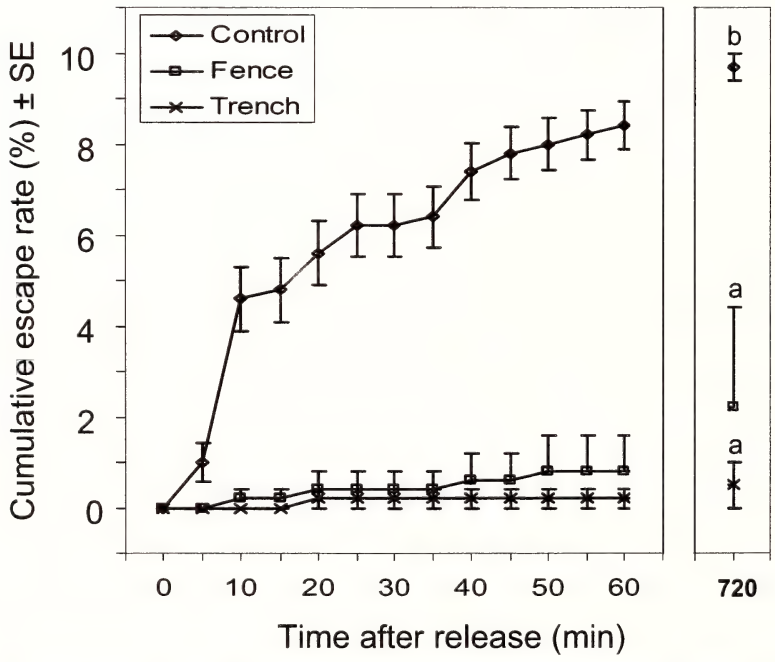

Figure 1. Black vine weevil escapes from a one m square area surrounded by a 20 cm high aluminum fence with fluoropolymer-coated tape attached inside (fence), a portable exclusion trench (trench) or no barrier (control). Observations were made at 5 min intervals for 1 h after insect release and 12 h after release. Lower error bars omitted from fence data points for clarity. Final means labeled with the same letter do not differ significantly at $\alpha = 0.05$ (Tukey's HSD test, $n = 3$).

Table 1.

Mean percentage of adult black vine weevils that left plastic canisters or cups that were untreated (control) or coated inside with dried fluoropolymer, white lithium grease (grease) or powdered talc (talc). Canisters and cups were placed in an open container (ambient RH) or a closed container with an open water source (saturated RH). Canisters were rinsed immediately before being placed in the closed container, leaving their surface wet.

	Canister escapes (%)[1], $n = 10$			Cup escapes (%)[1], $n = 9$		
Treatment	Dry surface, ambient RH	Wet surface, saturated air		Dry surface, ambient RH	Dry surface, saturated air	
Control	100 A	90 a A	$t_{18} = 2.3$, $P = 0.15$	89 a A	100 a A	$t_{16} = 1.0$, $P = 0.33$
Fluoro-polymer	0 A	10 b A	$t_{18} = 2.3$, $P = 0.15$	-	-	
Grease	0	0 b		0 b A	33 b A	$t_{16} = 4.0$, $P = 0.06$
Talc	0 B	70 a A	$t_{18} = 73$, $P < 0.0001$	0 b	0 c	
		$F_{3,39} = 50$		$F_{2,24} = 64$	$F_{2,24} = 38$	
		$P < 0.0001$		$P < 0.0001$	$P < 0.0001$	

[1] Means followed by the same lower case letter within a column do not differ significantly (Tukey's test, a= 0.05); those followed by the same upper case letter within a study and row do not differ significantly (t-test, a= 0.05).

escape. Weevils in fluoropolymer-treated canisters largely ceased their activity until the experiment ended. No weevils escaped from moistened grease-treated canisters.

Plastic surface treatments (saturated vs. ambient RH). Visible condensation first appeared on cups in the saturated environment 8 h after the test began and was very heavy by the end of the test. No condensation was seen on cups in the lower humidity environment. Almost all weevils escaped from control cups within the first hour of observation; the only weevil that did not escape from a control cup in an hour did not escape at all (Table 1). No weevils escaped from cups treated with talc in either container. One third of the weevils escaped from grease-treated cups in the saturated environment, but none escaped in the ambient RH environment (Table 1). On average, escapes from greased cups took longer than escapes from untreated cups in the sealed container (16.7 ± 0.7 versus 0.7 ± 0.7 h, respectively; $\chi^2 = 27$, df = 2; $P < 0.001$). Mean escape times from untreated cups did not differ between the open and sealed containers.

DISCUSSION

Under dry conditions talc dust, fluoropolymers and lithium grease treatments rendered several smooth surfaces (glass, plastic, and aluminum) unclimbable to BVW adults for the duration of our tests. Equivalent treatments were sometimes less effective under wet conditions, or in saturated environments. This may help explain why physical barriers that would be expected to offer total exclusion, based on observations under dry conditions, exclude only two-thirds of root weevils in the field (Bomford and Vernon 2005).

Most adult weevils quickly attempted to leave the open containers we used for our tests. Their success in exiting, and the length of time they took to leave, were considered indicators of the difficulty they had in scaling the barriers they faced. Under dry conditions, surface treatments eliminated escapes; under wet conditions they usually reduced the proportion of insects able to escape and lengthened escape times.

Cowles (1995) has suggested that root

weevils are able to evade physical barriers because natural bridges form over otherwise unclimbable surfaces. He has seen field debris, such as twigs, adhering to the white lithium grease on his barriers, and plant canopies touching across barriers (R.S. Cowles, pers. comm., see Acknowledgements). We have also seen natural bridges that could allow root weevils to cross portable trench barriers in field studies (Bomford and Vernon 2005), but these were not a factor in the tests reported here.

We observed repeated instances of BVW adults crossing vertical surfaces treated with fluoropolymer, talc dust, and lithium grease in the presence of moisture. BVW adults scaled talc-dusted plastic that had been lightly rinsed to mimic rainfall on a dusted plastic exclusion trench. Similar observations have been reported previously for Colorado potato beetles challenged by plastic-lined trenches after rainfall in field studies (Boiteau *et al.* 1994). Rinsing did not render greased surfaces climbable in one test, reflecting field observations in which greased aluminum barriers excluded root weevils after irrigation (Cowles 1995). We did, however, observe BVW scaling greased plastic with visible surface condensation in a high humidity environment and scaling fluoropolymer-treated aluminum in a light rain shower. We are unaware of other reports of moisture enhancing an insect's ability to scale fluoropolymer or lithium grease-coated surfaces. These observations lead us to suggest that the insects' tarsal pads adhere to condensation on treated surfaces. Essentially we hypothesize that the insects can overcome physical barriers by walking on water.

More rigorous tests of this hypothesis are necessary. The studies reported here reflect a variety of treatment combinations observed under different conditions. Experimental factors were sometimes confounded. For example, BVW were unable to scale a fluoropolymer treated fence under dry conditions two and seven days after the fence was erected, but scaled the same fence in a light rain shower nine days after setup. We attributed this difference to the presence of moisture, but it might also have been an effect of fence age. Similarly, our analyses of interactions between surface treatment and environment were confounded by the fact that surface treatments were replicated within environments, but only one instance of each environment was tested in any study. Our observations suggest intriguing avenues for further study, not definitive conclusions.

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Sources of Spring and Fall Hop Aphid, *Phorodon humuli* (Schrank), (Homoptera: Aphididae) Migrants in South Central Washington

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ABSTRACT

The hop aphid, *Phorodon humuli* (Schrank), flies from hop, *Humulus lupulus* L., to its overwintering *Prunus* spp. hosts in the fall. The sources of these aphids were not known because much of the aphid flight occurs after hop plants are removed from fields during harvest. We found that the bottoms of hop plants remaining alive in harvested hop yards averaged 1.7 to 5.8 hop aphids per leaf in three years of sampling. Unharvested hop plants remaining after harvest averaged 32.8 to 127.1 aphids per leaf in two years. Feral hops were also infested with hop aphids in late summer and early fall. Sources for the spring aphid flight from *Prunus* spp. to hop included *Prunus cerasifera* Ehrhart, which averaged 44.0 to 105.1 aphids per shoot in two years of sampling. Fruit-type *Prunus* spp. trees growing on residential properties averaged 0.9 and 11.3 aphids per shoot in the same years but few of these trees were found. Plum and prune orchards averaged 0 to 5.5 aphids per shoot in two years and estimates indicate that orchard trees are much more numerous than other hop aphid host trees. Potential alternative management strategies for hop aphid control are discussed.

Key Words: Homoptera, *Phorodon humuli*, hop, *Humulus lupulus*, *Prunus*, host plants

INTRODUCTION

The hop aphid, *Phorodon humuli* (Schrank), is an important pest of hop, *Humulus lupulus* L., in south central Washington state (WA) and in most hop-growing areas of the Northern Hemisphere (Neve 1991). It is a holocyclic aphid that overwinters in the egg stage on purple-leafed ornamental flowering plum, *Prunus cerasifera* Ehrhart (also known as cherry plum or Myrobalan plum), *Prunus divaricata* Ledebour, *Prunus domestica* L., *Prunus insititia* L., *Prunus mahaleb* L., and *Prunus spinosa* L. (Blackman and Eastop 1994). Eggs hatch in February and March followed by a variable number of generations of parthenogenetic wingless females. The winged females that fly to hop appear in WA in early to mid-May and flight continues from mid-July to early August (Wright *et al.* 1995). Hop is the aphid's only secondary (summer) host (Born 1968; Miciński and Ruszkiewicz 1974; Eppler 1986). Parthenogenetic, wingless females are produced on hop during the summer (Campbell 1985; Campbell and Tregidga 2005). In late August, gynoparae (winged females) are produced on hop, which begin the flight back to *Prunus* spp. Winged males that fly from hop to *Prunus* spp. appear about mid-September. Aphid flight often continues into November and is terminated by foliage-killing frost (Wright *et al.* 1995). The gynoparae give birth to a generation of wingless females, the oviparae, which mate with winged males and lay the overwintering eggs on *Prunus* spp. buds and stems. Neither hop aphids nor their eggs have been reported on hop during the winter. Further-

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more, gynoparae do not settle on hop leaves or reproduce on hop (Campbell and Tregidga 2005).

The aerial parts of hop plants are killed by fall frosts and only the hop roots, which are several cm below the soil surface, survive the winter. In the spring, shoots grow from the roots and they are trained to grow up fiber strings which are tied to a trellis that is about 5 m tall. During harvest (mid-August to mid-September) hop plants are cut at the top of the trellis and about 1 m above ground, removed from the fields and taken to stationary picking machines where the cones are separated from the leaves and stems. The cones are dried in large kilns at 60 °C and the waste leaves and stems are chopped and spread on the fields soon after harvest or after being stored in large piles. It is considered unlikely that many aphids could survive the picking process (Campbell and Tregidga 2005). Following harvest, about 1 m of basal foliage remains alive in hop fields until it is killed by frost. The amount of foliage remaining is quite variable ranging from a few leaves to hundreds of leaves per plant. Intact plants growing up trellis poles remain in some hop yards following harvest and feral (wild) hop plants are also present in the hop-growing region of WA (James *et al.* 2001). Approximately half of the gynoparae and very few males have flown by the end of harvest (Wright *et al.* 1995). One of our objectives was to determine if harvested and unharvested hop plants remaining alive in the fields after harvest, as well as feral hop plants, could be a source of fall migrants.

Another objective was to determine the source of aphids that fly from *Prunus* spp. to hop in the spring and summer. The hop-growing area of Washington is an area of diverse agriculture including a small number of plum or prune, *Prunus domestica* L., orchards. In addition, landowners have planted ornamental and fruit *Prunus* spp. near residences, businesses, and in parks. Determining the sources of the spring and fall migrants not only adds to our knowledge of the aphid's life cycle but also may reveal alternatives to the traditional control methods that are used on hops during the growing season.

MATERIALS AND METHODS

Aphids in harvested hop yards. Hop yards selected for sampling in three years (1984, 1987, 1989) were in the Prosser - Grandview area of the Yakima Valley, WA. In 1984, plants in 11 harvested hop yards were sampled between 25 September and 19 October. Apterae were identified in all field studies described in this manuscript with the aid of a 10X hand lens and the descriptions in Blackman and Eastop (1984). Hop aphids were counted in the field on one leaf per plant from each of 200 randomly selected plants in eight hop yards and from 100 plants in three yards. One leaf was sampled from each of 100 randomly selected plants per yard: in 27 hop yards (one yard had 94 samples) from 25 September to 6 October, 1987; and in 33 hop yards (one yard had 89 samples) from 15 September to 9 October, 1989.

A small number of hop yards had varying numbers of unharvested, intact hop plants growing up the trellis poles. Six to 100 (mean = 43.7) randomly selected unharvested plants were sampled in each of 11 yards between 25 September and 7 October, 1987 and 11 to 58 (mean = 27.2) unharvested plants were sampled in each of nine yards from 15 to 29 September 1989. One leaf from about the 2 m height, which is a representative sample (Wright *et al.*1990), was sampled per plant. The varieties sampled in all years were Cascade, L1 (Cluster), and Galena.

The mean aphids per leaf on harvested plants was compared with the mean per leaf on unharvested pole plants using the non-parametric Wilcoxon Rank Sum test computed by the NPAR1WAY procedure of SAS (SAS Institute 1988).

Aphids on feral hop plants. Six sites with feral hop plants were located in the

Yakima Valley of south central WA (James *et al.* 2001). The plants grew on fences, or poles, usually near roads. In 1999, the plants were sampled on 7 to 8 September and 11 to 12 October and in 2000, on 14 to 22 August and 18 to 19 September. Thirty leaves were collected randomly per site and the number of aphids per leaf were counted under a stereomicroscope in the laboratory.

Survey of hop aphids on *Prunus* in the spring. The survey area was divided into two adjacent hop growing areas of WA: one in western Benton County near Prosser, and the other in eastern Yakima County near Sunnyside, Grandview, and Mabton. Each area was about 15,540 ha. Surveys were conducted in 1990 (18 to 26 June) and 1991 (25 June to 5 July). In 1990 we drove the roads in an unsystematic pattern and located *P. cerasifera* and fruit varieties of *P. domestica* by sight. Orchards were sampled by selecting 10 trees at random and sampling 10 shoots per tree. Hop aphids in spring are concentrated on the new foliage near the tips of the shoots (Wright *et al.* 1995). In addition to the hop aphid, we found the mealy plum aphid, *Hyalopterus pruni* (Geoffroy), and the leaf-curling plum aphid, *Brachycaudus helichrysi* (Kaltenbach). Ornamental and fruit trees at residences and commercial properties that were not orchards were sampled by examining 10 shoots per tree or shrub. Some small trees did not have 10 shoots, so fewer shoots were sampled on those trees. Aphid numbers were expressed as the number per shoot. Usually every tree at a site was sampled but if a property had more than three or four trees, a subsample of trees was selected. In 1991, the survey was done systematically. Most of the roads in the surveyed area are laid out in a grid of squares that are 1.6 km on a side. Road sections of 1.6 km each were selected at random on a map and 14 % of the roads in each area were surveyed as in 1990. For orchards, the number of trees per ha was calculated by multiplying the number of orchards in the surveyed area by 1,272, which was the average number of trees per plum and prune farm in Benton and Yakima counties (the counties of hop production) in 1992 (National Agricultural Statistics Service 1992) and dividing by the area surveyed. The number of trees not in orchards was determined by dividing the number of trees in the survey by the hectares in the area surveyed.

RESULTS

Aphids in harvested hop yards. We found hop aphids on the bases of harvested hop plants and on unharvested plants growing on trellis poles (Table 1). The unharvested plants had significantly more aphids per leaf than the harvested plants. Only two yards in the three years of sampling had no aphids in the samples.

Aphids on feral hop plants. In 1999, we found a mean of 0.7 aphids per leaf on 7 to 8 September (range = 0 to1.6) and 20.9 on 11 to 12 October (range = 0 to 93.6). In 2000, there was a mean of 0.7 per leaf (range = 0 to 1.7) on 14 to 22 August and 11.7 (range = 0 to 30.3) on 18 to 19 September.

Survey of aphids on *Prunus* in the spring. In 1990, 14 commercial prune orchards were sampled and hop aphids were found in four of them. The mean number of aphids per shoot in all orchards was 5.5 but most of the aphids were found in one orchard that averaged 81.0 aphids per shoot. Fruit-type *Prunus* were found at three residences with one tree each and aphids were found on two of the trees. The mean from all three trees was 0.9 aphids per shoot. Seventy-two purple-leafed ornamental plum trees were sampled at 42 sites and hop aphids were found on 50 trees at 32 sites. The number of trees sampled per site ranged from one to eight. The mean number of hop aphids on all ornamental trees was 44.0 per shoot.

In 1991, we found four commercial prune orchards and no hop aphids were found in any of them. A total of seven fruit-type plums were found at five residences

Table 1.
Mean number of hop aphids per leaf on harvested and unharvested hop plants remaining in hop yards in September and October. N, total number of leaves sampled (one leaf per plant). Z, test statistic for Wilcoxon Rank Sum test.

Year	Plant type	Mean aphids (range)	N	Z
1984	harvested	4.4 (0.2 – 13.2)	1,900	na
1987	harvested	1.7 (0 – 18.9)	2,694	8.2[1]
	unharvested	32.8 (0.2 – 316.5)	481	
1989	harvested	5.8 (0 – 56.4)	3,289	14.8[1]
	unharvested	127.1 (0.2 – 481.6)	245	

[1] $P<0.0001$.

but aphids were found on only two trees at one site with an average of 39.5 aphids per shoot. The mean for all fruit trees at residences was 11.3 aphids per shoot. We sampled 57 purple-leafed ornamental plum trees at 37 sites and hop aphids were found on 36 trees at 27 sites. The mean number of hop aphids on all trees was 105.1 per shoot. The estimated number of trees per ha was 1.16 for orchard trees, 0.017 for purple-leaf ornamental flowering trees and 0.0016 for fruit trees at residences.

DISCUSSION

Hop aphids were common in harvested hop yards, indicating that harvested hop yards were a major source of the aphids for the fall flight to *Prunus*. Hop plants growing up the trellis poles had more leaves than the bottoms of harvested plants and were infested with more aphids per leaf (Table 1); however, unharvested plants were uncommon compared to the number of harvested plants, so they probably contribute a small proportion of the hop aphids produced over the whole area.

Feral hop plants were infested with hop aphids, occasionally with high numbers. Hop is not native to the Pacific Northwest (Hitchcock and Cronquist 1973) and only female plants that produce seedless hop cones are grown commercially in WA. These factors may restrict the number of feral hops growing in south central WA. Wild hops may be an important source of fall migrants in England (Campbell and Tregidga 2005). Our observations indicate that feral hops in south central Washington are scarce compared to the number of commercial hop plants but a more intensive survey would be needed to determine the population size of feral hops.

Our survey of *Prunus* spp. indicates that purple-leafed ornamental flowering plums were a major source of spring migrant hop aphids. Only one commercial prune orchard was heavily infested with hop aphids but, because of the large number of trees in this orchard, it could be a significant source of aphids. Orchard trees are usually sprayed with insecticides to control aphids and this is probably the main reason aphid numbers were generally low in orchards. Since this survey was done, the plum and prune industry has declined from 565 ha in Benton and Yakima counties in 1992 to 311 ha in 2002 (National Agricultural Statistics Service 1992; 2002). The ornamental varieties were much less abundant than orchard trees but they were infested with higher densities and they were well dispersed throughout the survey area.

Knowing the sources of the spring and fall migrating aphids and the timing of the flights suggests some alternative aphid controls. As gynoparae start flying before harvest is completed and males start flying near the end of harvest in mid to late Sep-

tember (Wright *et al.* 1995), controlling aphids in harvested hop yards would reduce the number of gynoparae but should be more effective in reducing the number of males. The desired result would be a reduction in the number of mated females and eggs on *Prunus* spp. Potential control of aphids in harvested hop yards could involve insecticide applications, destroying the foliage with cultivation, or defoliation with herbicides. Because unharvested plants contribute nothing to the harvest, permanently removing them or cutting them off at the base during harvest would be a good field sanitation practice. A potential secondary problem may be the disruption of insect and mite natural enemies in hop yards (Strong and Croft 1993; James *et al.* 2001).

Successful control of hop aphids on harvested hops would depend on hop growers over a large area cooperating in a fall control program. Controls would have to be applied as soon after harvest as possible and would need to be extremely effective. Workers in Idaho developed an area-wide program to reduce potato leaf roll virus by reducing the number of green peach aphids, *Myzus persicae* (Sulzer), in the spring before the aphids flew to potatoes (Bishop 1967). They sprayed insecticides on introduced flower and vegetable transplants and home gardens, and removed the aphid's overwintering hosts, peach and apricot trees. This program was successful in reducing aphids and potato leaf roll virus when spraying was thorough and well timed. The small size and isolation of the potato-growing areas were important factors in the program's success.

The hop-growing region of Washington is isolated from other hop-growing areas, so perhaps a similar area-wide program could be effective against the hop aphid. Controlling aphids in prune and plum orchards would be essential. For ornamental trees, one potential method would be the removal of *Prunus* spp. host trees, especially *P. cerasifera*. Dixon and Kindlmann (1990) present theoretical evidence that aphid abundance is directly related to host plant abundance and the number of hop aphids caught in suction traps in England and Washington is related to the abundance of host plants in the area (Taylor *et al.* 1979, Wright *et al.* 1995). This suggests that the hop aphid populations may be susceptible to manipulations of host plant abundance. Hymenopterous parasitoids commonly attack hop aphids on *Prunus* spp. in the spring (Wright and James 2001). Perhaps parasitoids and predators could be managed to reduce the number of spring migrants flying to hops. Spraying ornamental *Prunus* spp. may be effective but could have negative impacts on natural enemies. Because the hop aphid can migrate over long distances (Taylor *et al.*1979), any area-wide program would need to cover a large area to be effective. To be successful, any alternative control would have to provide significantly superior control, be safer to people or the environment, or cost less than traditional methods.

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Host preference by *Saperda calcarata* Say (Coleoptera: Cerambycidae)

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ABSTRACT

We conducted five laboratory and one field experiments to examine potential host selection mechanisms of *Saperda calcarata* Say in British Columbia. Olfactory bioassays indicated that female (and possibly male) beetles were attracted to volatiles from leafy twigs of trembling aspen, *Populus tremuloides* Michaux. However, wounding of the bole, ethanol baiting, or both, did not result in significant orientation toward or attack of trembling aspens in the field. Feeding preferences for trembling aspen were strong for both sexes in choice bioassays, but in no-choice bioassays, females did not discriminate between trembling aspen and black cottonwood, *P. trichocarpa* Torrey & Gray. Scouler's willow, *Salix scouleriana* Barrat in Hooker, was fed upon the least by both sexes. When diameter of bolts offered as oviposition hosts was equalized, frequency of oviposition was similar among the three hosts. Our data suggest that feeding preference is the predominant mechanism of host selection by *S. calcarata*.

INTRODUCTION

The poplar borer, *Saperda calcarata* Say (Coleoptera: Cerambycidae: Lamiinae) attacks living poplars from the sections Populus (*P. tremuloides* Michaux, *P. alba* L., *P. grandidentata* Michaux), Aigeiros (*P. deltoides* Bartram, *P. fremontii* Watson, *P. nigra* L. 'Italica'), and Tacamahaca (*P. angustifolia* James, *P. balsamifera* L., *P. trichocarpa* Torrey & Gray) throughout their range in North America (Hofer 1920, Baker 1972, Drouin & Wong 1975, Nebeker *et al.* 1985). The beetle also attacks poplar hybrids (*P.* x *acuminata*) (Hofer 1920) and willows (Baker 1972). *Populus* spp. are susceptible from approximately three years of age (Abrahamson & Newsome 1972), or 4-5 cm diameter at breast height (dbh = 1.3 m) (Hofer 1920, Drouin & Wong 1975, Nebeker *et al.* 1985). *Saperda calcarata* adults reportedly discriminate among poplar hybrids for feeding (Garland & Worden 1969) and there are differences in attack rates among *P. deltoides* clones (Nebeker *et al.* 1985).

In British Columbia (BC), *S. calcarata* adults emerge in late June to July, undergo a short period of maturation feeding (Linsley 1959) and mate. Females oviposit into oblong niches chewed in the bark of host trees. Young larvae mine in the inner bark and sapwood, then move deeper creating large, irregular galleries throughout the sapwood and heartwood (Hofer 1920). Frequently a single tree is repeatedly attacked forming a 'brood' tree. Attacked trees are identified by their deformed bole, oviposition scars, sap stains spreading down the bark, and frass piles at their base. The life cycle takes three to four years in Canada, but is probably shorter in the south (Hofer 1920, Peterson 1947, Baker 1972).

Saperda calcarata is considered a major pest of poplars (Solomon 1987) and frequently becomes prevalent within stands (Bird 1930, Nebeker *et al.* 1985). Physical damage to the boles from larval galleries makes trees susceptible to breakage. Openings from oviposition niches and woodpeckers lead to increased incidence of pathogens like *Hypoxylon mammatum* (Wahlenberg) Karsten (Graham & Harrison 1954) or *Phellinus tremulae* (Bondartsev)

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Bondartsev & Borisov (Hofer 1920) which girdle the bark or stain and rot the wood, and attacked trees may be further damaged by other insects, e.g., *Agrilus anxius* Gory or *Poecilonota cyanipes* (Say) (Hofer 1920).

Because attacks by beetles may be prevalent in poor sites, e.g., dry slopes (Hofer 1920, Bird 1930, Morris 1963), or in decadent hosts (Graham & Harrison 1954), *S. calcarata* is assumed to prefer weakened hosts that remain alive during attack (Hanks 1999). In agreement with this hypothesis, less attack was observed on *P. deltoides* clones from southern provenances which grew most vigorously (Nebeker *et al.* 1985). In contrast, Baker (1972) noted that brood trees were larger and faster growing than neighbouring trees, and Abrahamson & Newsome (1972) concluded there was no difference in attack level on different quality sites. Olfaction is generally believed to play a large role in cerambycid host location (Linsley 1961, Hanks 1999, Allison *et al.* 2004).

We commonly observe *S. calcarata* attack in trembling aspen, *P. tremuloides*, in BC, but not in black cottonwood, *P. trichocarpa*, or willow, *Salix* spp. Our objectives were to determine: if olfactory attraction occurs to trembling aspen, the apparent preferred host, and if there are different levels of feeding or oviposition among these three hosts.

MATERIALS AND METHODS

Saperda Colonies. We collected ca. 2.5 m^3 *S. calcarata*-attacked trembling aspen bolts from trees felled near 70 Mile House, BC in April or May of 2002 to 2004. Adults emerged from the caged bolts during June and July for two successive years. A total of 76 and 101, 46 and 30, and 11 beetles emerged each year from bolts harvested in 2002, 2003 and 2004, respectively. Timing of emergence was in agreement with Garland & Worden (1969). Adults were kept on aspen branches in water in 1.2 x 1.8 x 0.6 m outdoor enclosures until used in bioassays. Beetles were used once in any one type of bioassay, except for feeding bioassays in 2002, when tested beetles were returned to the holding cage from which test subjects were removed.

Plant material. Leafy aspen branches were collected periodically, mostly from various interior BC locations, but also from Burnaby and Maple Ridge on the coast. Branches were kept with the cut ends in water at 4 °C, and used in bioassays within one week. Both Scouler's willow, *Salix scouleriana* Barrat in Hooker, and black cottonwood branches were collected in Burnaby, BC the same day bioassays were performed. In total, four to five genotypes of each species were tested.

Olfaction experiments. In 2003 and 2004, responses to volatiles were investigated in the laboratory using a still air olfactometer (Figure 1). Randomly assigned treatment and control jars contained either a small jar of water with a small, leafy branch of aspen, or just water, respectively. The arena ceiling was a sheet of clear Lexan (GE Polymershapes, Coquitlam, BC). A video camera was positioned above the arena. To induce the photopositive beetles to approach the stimuli, a fluorescent light was placed under the platform between the two jars. The entire apparatus was covered with a black cloth. Three to five adults of a single sex taken directly from the holding cage were placed into each arena. Presence of beetles in the concentric rings above the treatment or control jars were determined at 30 sec intervals for 2 h. Fourteen trials were recorded. Feeding damage to the perforated centres above control and treatment stimuli was assessed for both recorded and unrecorded trials. Jars were washed and the paper covering replaced before each assay. Assays commenced any time between 0800 and 2400 h.

A field experiment was set up in a trembling aspen grove near Sabiston Lake, northeast of Savona, BC, on 28 June, 2002. Apparently healthy trees, spaced approximately 20 m apart, received one of four treatments in a randomized block design (n = 14): 4 axe cuts on opposite sides of the

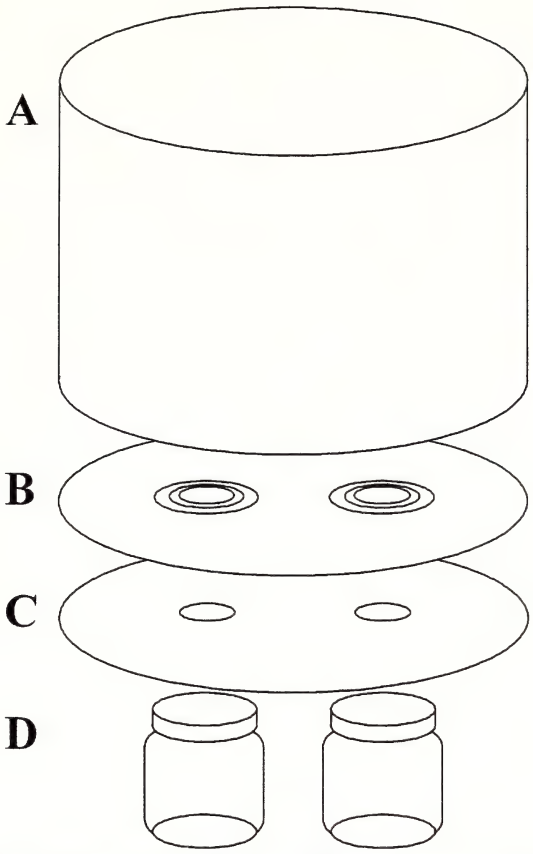

Figure 1. Schematic of olfactometer used to test olfactory responses of *S. calcarata* to trembling aspen leaf volatiles. A 60 cm diameter circular arena consisted of a black cylinder 60 cm high (A) placed on a white coroplast (GE Polymershapes, Coquitlam, BC) platform (C) with two holes, 5.5 cm in diameter with centres 28 cm apart. This platform was covered with white paper (B) perforated 41 times with a pin in a uniform, radial pattern above the openings. Three concentric rings 5.5, 12.5, and 19.5 cm diameter were drawn on the paper (B) above each hole. The platform (C) rested on glass cookie jars (aperture 11 cm diameter) (D), centred below each hole.

tree at ca. 1.5 m; ethanol bait stapled at ca. 2 m; 4 axe cuts plus ethanol bait; or no treatment. Ethanol is a ubiquitous kairomonal indicator of stressed trees (Kelsey & Joseph 1998 and references therein). The basal 3 m of the trees were examined for oviposition in 2003 and 2004.

Feeding bioassays. Choice and no-choice feeding bioassay experiments were performed to investigate feeding preferences of *S. calcarata* among trembling aspen, black cottonwood and Scouler's willow. Three small branches, each with three to five leaves, were placed in water-filled vials inside 17x16x12 cm plexiglass boxes. A single *S. calcarata* adult was allowed to freely feed on plant material overnight. No-choice bioassays contained one of the three potential hosts, and choice bioassays contained one branch of each. Before an assay, the leaves were traced onto paper. After the bioassay ended, leaves were attached to their traced counterparts, scanned, and the leaf area consumed was quantified using Scion Image software (Scion Corporation, Frederick, Maryland). A total of 17 choice and 14 no-choice bioassays were performed during July of 2002 and 2003.

Oviposition bioassays. In 2002 we tested oviposition by *S. calcarata* in holding cages on eight freshly cut bolts of varying diameter of each of the above three species. In 2003, two apparently healthy trembling aspen, black cottonwood, and

Scouler's willow trees of similar diameter were felled on 1 and 18 July 2003, bucked and transported to SFU where they were kept refrigerated until needed. Six to eight beetle pairs were placed in 13 outdoor cages, 90 x 90 x 90 cm, with one randomly positioned bolt of each species and a central water jar containing leafy trembling aspen branches for seven days. Bolts were ca. 50 cm in length, and diameters taken from their midpoint. Oviposition was determined in both experiments by opening all niches cut in the bark.

Statistical analyses. In all cases α = 0.05. For olfaction bioassays, one-tailed paired t-tests were used to determine if the frequency of observations in each of the concentric rings was greater over treatment than control jars. Chi-square tests were performed for each sex to compare the frequency of feeding damage above treatment *vs.* control stimuli against the null hypothesis of no discrimination between stimuli. Data from feeding and oviposition bioassays were transformed by $x^{1/2}$ and log(x+1), respectively, to correct for non-normality and heteroskedasticity, then analyzed as randomized complete blocks by ANOVA with PROC GLM (SAS Institute 1990). Because it was not possible to perform all no-choice feeding assays at the same time, the analysis included trial date and host species effects. The final model did not include interaction effects. Multiple comparisons were performed with REGWQ (SAS Institute 1990).

RESULTS

Olfaction experiments. There were no differences in the occurrence of beetles in the middle and outside rings bordering the perforated area above treatment (i.e. trembling aspen leaves) and control stimuli for both sexes (middle ring, females, t = 0.69, P = 0.26, males, t = -0.80, P = 0.23; outer ring, females, t = 0.76, P = 0.24, males, t = 0.95, P = 0.19). Females (but not males) were present more frequently above the treatment than the control stimulus (Table 1). Often, females and males fed on the perforated paper directly above the treatment stimulus, but in one instance females fed above the control as well (Table 1). Females also chewed curvilinear patterns in the paper covering the arena floor that were reminiscent of oviposition niches, but no eggs were found.

Very few attacks were found on trembling aspen treated to release host volatiles (axe cuts) or baited with ethanol. A total of 8 oviposition chambers in two replicates were found: 1 niche on an ethanol-treated tree; 1 and 3 niches on two axe-cut trees; 2 and 1 niches on two trees with both treatments and none on control trees. None of these niches developed into successful larval galleries.

Feeding bioassays. We observed feeding on both the petioles and leaves as did Garland & Worden (1969), but only quantified the more abundant foliar damage. When given a choice (Figure 2), both sexes clearly preferred trembling aspen over both black cottonwood and Scouler's willow (females $F_{2,32}$ = 41.15, P < 0.0001; males $F_{2,32}$ = 42.68, P < 0.0001). There were also significant differences in feeding in the no-choice experiment (females $F_{2,34}$ = 20.53, P < 0.0001; males $F_{2,33}$ = 26.96, P < 0.0001). However, females accepted trembling aspen and black cottonwood equally, and males fed on black cottonwood more vigorously than on Scouler's willow (Figure 2).

Oviposition bioassays. When bolt diameter was not controlled in 2002, *S. calcarata* females oviposited preferentially in trembling aspen and black cottonwood, the species with the largest diameter bolts (Table 2). When bolt diameter was equalized in 2003, there was no preference in oviposition among the three host species (Table 2).

We observed some larvae feeding in the bark of all three species, but they did not survive long as bark quality deteriorated rapidly because of infection by *Cytospera chrysosperma* (Persoon: Fries) Fries, distinguished by characteristic orange conidial tendrils (Callan 1998).

Table 1.
Comparison of behavioural activity by male and female *S. calcarata* within perforated centres of arena floor above treatment (i.e. trembling aspen leaves) and control jars in the still-air olfactometer.

Observations	Females	Males
Observations of beetles within arena circle circumscribing perforated area above treatment or control stimulus, 30 sec intervals for 2 h.		
no. replicates	7	7
mean no. observations ± SE		
treatment stimulus	55.4 ± 18.5	41.9 ± 8.8
control stimulus	26.3 ± 10.8	41.9 ± 18.5
t-value	1.99	0.00
Probability	0.047	0.50
Observations of feeding on perforated area of arena floor above treatment or control stimulus		
no. replicates	8	11
no. times most feeding above treatment stimulus	7[1]	6
no. times most feeding above control stimulus	0	0
no. times no feeding damage observed	1	5
Chi-square value	7.00	6.00
Probability	0.008	0.01

[1] In one trial feeding damage was observed above both stimuli, but damage was much greater over the treatment stimulus.

DISCUSSION

Our results indicate that *S. calcarata* can locate potential hosts by olfaction, can discriminate among tree species through gustatory cues, and may reject trees for oviposition if their diameter is too small.

Females were attracted to the volatiles from leaf-bearing twigs of trembling aspen in an arena olfactometer (Table 1). The fact that males chewed the paper above treatment but not control stimuli suggests they too are attracted to host volatiles. In the field experiment, we had hypothesized that *S. calcarata* brood trees would produce ethanol, and possibly other metabolites caused by wounding, and that the combination would be attractive. The positive response to leafy twigs, and the failure to induce significant attack on trees that were wounded, ethanol-baited, or both, suggests that initial orientation is to volatiles from leaves on which adults feed. Oviposition tends to be located in the upper parts of the bole beneath the canopy (Peterson 1947), requiring little movement by feeding beetles. Similarly, in an unpublished study conducted by C.L. Broberg and R. Gries (SFU), 15 antennally-active volatiles from the bark of trembling aspen were identified by coupled gas chromatographic electroantennographic detection analysis. However, in six field-trapping experiments, no *S. calcarata* were captured to various partial or complete blends of these compounds. These results may indicate that *S. calcarata* use leaf volatiles in orientation toward suitable hosts during flight.

The lack of a strong olfactory response by *S. calcarata* is not surprising. As a specialist of weakened hosts that may support multiple generations on the same tree (Hanks 1999), *S. calcarata* is not likely subject to strong selection pressure to adapt to finding new hosts. Furthermore, *Populus* spp. are pioneer species and often occur in locally abundant populations. Thus emergent *S. calcarata* may not need to disperse

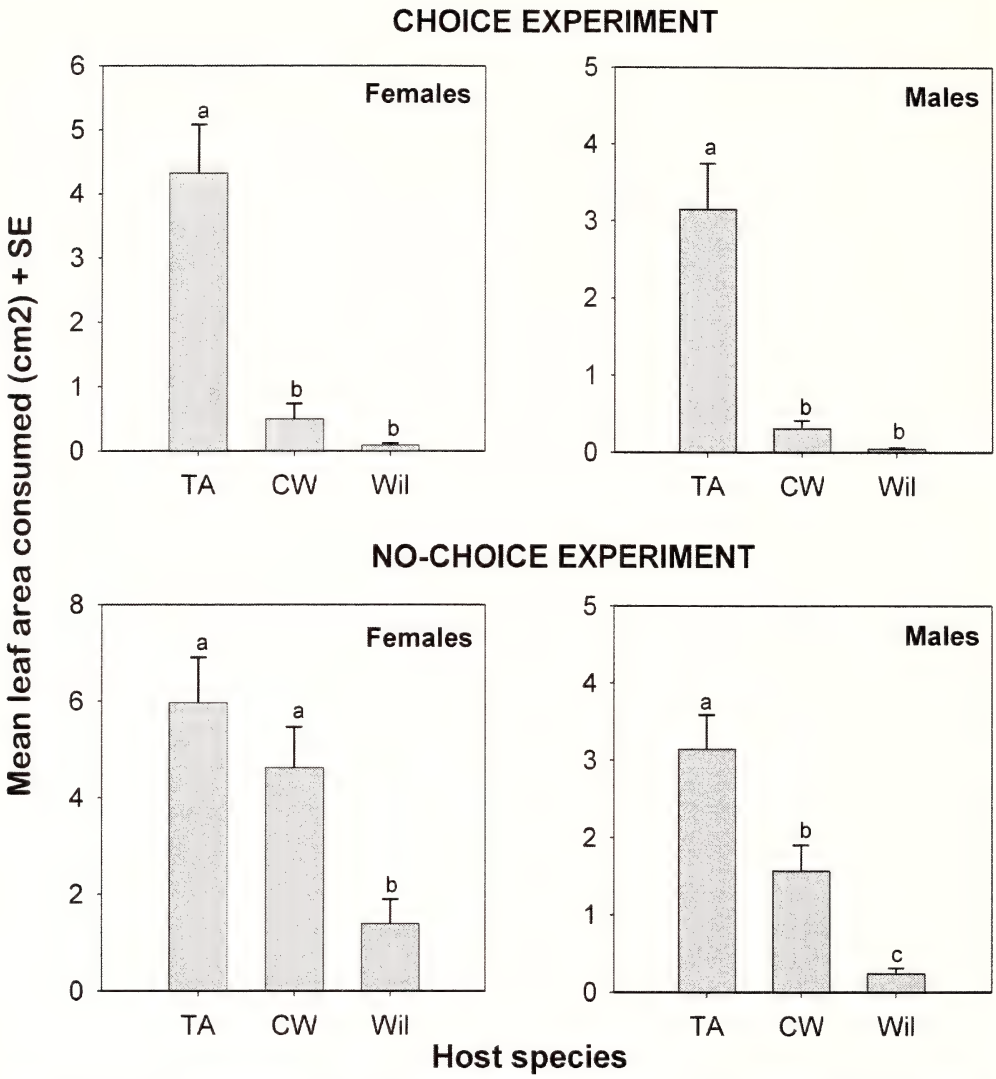

Figure 2. Leaf area consumed by female and male *S. calcarata* when presented with the three potential hosts simultaneously (choice experiment) or separately (no-choice experiment). Bars within an experiment and sex with the same letter are not significantly different, REGWQ test, $P < 0.05$. TA = trembling aspen, CW = black cottonwood, Wil = Scouler's willow.

long distances to find a suitable host. In contrast, stressed hosts which are moribund and can only support one generation of beetle (Hanks 1999) are often rare and/or patchy in distribution. Cerambycid specialists on these hosts have evolved strong long-distance response mechanisms that often involve orientation to host volatiles from recently downed or injured trees, smoke volatiles from burned trees, or pheromones produced by secondary bark beetles pheromones (Allison *et al.* 2004). Although these cerambycids mate and oviposit on these newly found hosts, they engage in maturation feeding on healthy trees (Hanks 1999). Weakened host specialists like *S. calcarata* can use a single individual for all functions; thus even the malformed emergent adults incapable of flight observed by us and others (Peterson 1947; Drouin & Wong 1975), can experience reproductive success without long range dispersal and olfactory orientation to suitable hosts.

Both sexes clearly preferred trembling aspen in choice feeding bioassays and re-

Table 2.
Comparison of oviposition by *S. calcarata* on bolts from three different hosts when bolt diameters were unequal or similar.

Experimental description	Species	Bolt diameter (cm)		Mean no. ovipositions ± SE [1]
		Range	Mean ± SE[1]	
Bolt diameters unequal (2002)	Trembling aspen	6.3 - 13.2	9.5 ± 0.8 a	21.0 ± 9.6 b
	Black cottonwood	8.9 - 16.7	12.2 ± 0.9 a	27.5 ± 9.4 a
	Scouler's willow	3.2 - 5.5	4.6 ± 0.3 b	0.4 ± 0.2 c
Bolt diameters similar (2003)	Trembling aspen	7.8 - 17.1	13.5 ± 0.6 a	4.4 ± 2.1 a
	Black cottonwood	8.1 - 17.3	13.1 ± 0.6 a	8.0 ± 3.2 a
	Scouler's willow	9.2 - 17.0	12.6 ± 0.6 a	8.3 ± 2.8 a

[1] Means within an experiment and column followed by the same letter are not significantly different, REGWQ test, $P < 0.05$. ANOVA statistics as follows: 2002 bolt diameter $F_{2,18} = 33.83$, $P < 0.0001$; oviposition $F_{2,18} = 21.17$, $P < 0.0001$; 2003 bolt diameter $F_{2,24} = 2.57$, $P = 0.10$; oviposition $F_{2,24} = 1.17$, $P = 0.33$.

jected other species, but in the no-choice bioassays, females did not discriminate between trembling aspen and black cottonwood, and males accepted black cottonwood more than Scouler's willow (Figure 2). Females may have higher nutritive requirements than males and therefore cannot afford to be as selective. Preference for trembling aspen could be a result of local adaptation to a species that comprises 4.6 times more wood volume in BC than black cottonwood (BC Ministry of Forests 1998). *Saperda calcarata* is mobile enough to sample numerous trees before finding one that is suitable for feeding.

There are few records in BC of *S. calcarata* attack on *Salix* spp. In Saskatchewan, however, both *Salix* and *Populus* spp. were reported to be "readily eaten" by adults (Peterson 1947). Thus, there could be host-related ecotypes in different regions of North America.

The lack of discrimination between hosts for oviposition when trembling aspen leaves were available for maturation feeding, and diameters of bolts from the three species were equalized, indicates that host volume is more important than species for larval feeding and development (Table 2). Oviposition in large-diameter hosts would be adaptive in ensuring that most hosts did not suffer breakage during the three to four years required for larval development.

In conclusion, lack of evidence for long-range olfactory orientation to new hosts, correlation between *S. calcarata* incidence in BC and gustatory preferences, high mobility, and lack of discrimination between hosts for oviposition, suggest that feeding preference constitutes the predominant mechanism of host selection by *S. calcarata* in BC.

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Yellowjacket Wasps (Hymenoptera: Vespidae) Trapped in Alaska with Heptyl Butyrate, Acetic Acid and Isobutanol

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ABSTRACT

Eight species of vespine wasps were captured in traps near Fairbanks, Delta Junction and Palmer, Alaska, during 2003 and 2004. These were *Vespula vulgaris* L., *V. acadica* (Sladen), *V. consobrina* (Saussure), *V. rufa* (L.)(=*intermedia* [Buysson]), *Dolichovespula maculata* (L.), *D. arenaria* (F.), *D. norwegica* (F.)(=*albida* [Sladen]), and *D. norvegicoides* (Sladen). Workers and males of *V. vulgaris* were captured primarily in traps baited with the combination of acetic acid and isobutanol. Workers of *V. acadica*, *V. consobrina*, and *V. rufa* were captured primarily in traps baited with heptyl butyrate. Queens and workers of *D. maculata* were captured primarily in traps baited with acetic acid, or acetic acid plus isobutanol. The small numbers of *D. arenaria*, *D. norvegicoides*, and *D. norwegica* captured did not permit treatment comparisons. Season-long trapping indicated a presence of *V. acadica, V. consobrina, and V. rufa* workers from late June through July, *D. maculata* from early July into early August, and *V. vulgaris* from late July to early September. The earliest wasps captured were queens of *V. vulgaris* and *D. maculata* in late May, while the latest wasp captured was a worker of *V. vulgaris* the first week of October, in Palmer.

Key Words: social wasps, Vespinae, *Vespula*, *Dolichovespula*, trapping, attractant

INTRODUCTION

There is little information available on the abundance, distribution, or seasonality of social wasps (Vespidae, Vespinae) in Alaska, despite their likely widespread and recurring pest status. Many species of yellowjackets, which belong to the genera *Vespula* and *Dolichovespula* (Greene and Caron 1980), are often stinging hazards to people, pets, and livestock. An early report from the Harriman Alaska Expedition (Kincaid 1900) listed only two species: *Dolichovespula norwegica* (F.) (as *Vespa marginata* Kirby) from Kukak Bay, and *D. arenaria* (F.) (as *Vespa borealis* Kirby) collected in Sitka. Distributions given for vespid wasps of North America by Miller (1961), Wagner (1978), Akre *et al.* (1980), Eck (1984), and Carpenter and Kojima (1997) indicate that *D. norwegica* (= *albida* Sladen), *D. adulterina* (Buysson) (= *arctica* Rohwer), *D. arenaria*, *D. norvegicoides* (Sladen), *D. alpicola* Wagner, *D. maculata* (L.), *Vespula vulgaris* (L.), *V. acadica (Sladen), V. austriaca* (Panzer), and *V. rufa* L. [= *intermedia* (Buysson)] are present in Alaska. The pest status of *V. vulgaris*, the common yellowjacket, in Alaska is indicated by Shippey (1994) (cited in Barnes *et al.* 1996).

Chemical attractants useful in trapping and monitoring yellowjacket wasps include heptyl butyrate (Davis *et al.* 1969), and acetic acid plus isobutanol (Landolt 1998). Heptyl butyrate is a strong attractant for *Vespula pensylvanica* (Saussure) (Davis *et al.* 1973), and also attracts significant numbers of *V. squamosa* (Drury) and some members of the *V. rufa* species group: *V. atropilosa* (Sladen), *V. acadica*, *V. consobrina* (Saussure), and *V. vidua* (Saussure) (Grothaus *et al.* 1973, MacDonald *et al.* 1973, Howell *et al.* 1974, Reed and Landolt

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2002, Landolt *et al.* 2003). Acetic acid plus isobutanol is attractive to some members of the *V. vulgaris* species group: *V. germanica* (F.), *V. pensylvanica*, *V. vulgaris*, and *V. maculifrons* (Buysson), as well as to *V. squamosa*, *Vespa crabro* L. and several species of *Polistes* (Landolt 1999; Landolt *et al.* 2001). Acetic acid or isobutanol alone are also weakly attractive to some species of social wasps (Landolt *et al.* 1999, Reed and Landolt 2002), and acetic acid was co-attractive with heptyl butyrate for trapping *V. pensylvanica* (Landolt 1998).

In this study, we sought to determine and compare the responses of species of social wasps in Alaska to heptyl butyrate, acetic acid, and isobutanol, particularly to test the hypothesis that yellowjackets in the genus *Dolichovespula* and the *V. vulgaris* species group are primarily attracted to acetic acid and isobutanol, while yellowjackets in the *V. rufa* species group are primarily attracted to heptyl butyrate. We also sought to confirm information on the species of social wasps that are present in Alaska and determine the seasonal pattern of abundance of species that are likely to be pestiferous. We report here the results of trapping tests that provide significant information on yellowjacket wasp responses to chemical lures, and on the seasonality of several species of Vespinae in Alaska.

MATERIALS AND METHODS

Dome or Trappitt® traps (Gempler's, Belleville, Wisconsin, USA) were used to capture attracted wasps. These traps are pear-shaped with clear plastic tops and opaque yellow bottoms within which is placed a drowning solution. Wasps enter the trap through the invaginated bottom of the trap. Attractants were dispensed from polypropylene vials with holes in the lid for chemical release. Each vial contained chemical attractant on cotton balls. Vial sizes, active ingredient load amounts, and hole diameters selected were based on results of previous studies of wasp responses, as well as chemical release rates from vials under laboratory conditions. At ambient laboratory temperature (22.5 °C), estimated rate of release of compounds from vials with 3 mm diam holes is 200 ug heptyl butyrate per hour (Landolt *et al.* 2003), 8.2 mg acetic acid per hour (Landolt and Alfaro 2001), and 10 mg isobutanol per hour (using gravimetric methods reported in Landolt and Alfaro 2001). Vials were suspended at the top of the inside of the trap. Traps also contained 200 to 300 ml of a drowning solution which was 0.125% unscented detergent and 2% boric acid in water. Traps were placed a minimum of 20 m apart, and were placed at a height of 1.0 to 1.5 m on vegetation or on fences. Traps were checked once per week, at which time the drowning solution was replaced, and lures were replaced every month, which would be before the attractant in the dispenser was depleted.

2003 Trapping Test. Five sets of traps were placed on the campus of the University of Alaska, Fairbanks North Star Borough, Alaska, during the second week of July, 2003. Trap treatments at each location were: 1) an unbaited trap as a control, and traps baited with 2) acetic acid, 3) isobutanol, 4) acetic acid plus isobutanol, 5) heptyl butyrate, and 6) acetic acid plus heptyl butyrate. Each chemical (10 ml load) was dispensed from its own 15 ml vial with a 3 mm diam hole. Trap sites were in the vicinity of forested tracts, agricultural land, and a horticultural garden.

2004 Trapping Test. Traps were set up in early May at three locations, as pairs of traps baited with two chemical attractants: heptyl butyrate and acetic acid plus isobutanol. Heptyl butyrate (10 ml load) was dispensed from a 15 ml vial with a 3 mm hole and acetic acid plus isobutanol was provided as a mixture of the two compounds (10 ml load) in a single 15 ml vial with a 6 mm diameter hole. Four pairs of traps were placed on the main campus of the University of Alaska, Fairbanks, five pairs of traps were placed at the University of Alaska field site at Delta Junction,

Southeast Fairbanks Borough, and four pairs of traps were placed at the University of Alaska field site at Palmer, Matanuska-Susitna Borough. Trap sites were near both forested and agricultural lands. Traps at Fairbanks and Palmer were maintained until the third week of September and traps at Delta Junction were maintained until the first week of October.

Insects captured in traps were placed in pre-labeled plastic locking freezer bags. A separate bag was used for each trap and for each day the trap was checked. These were stored in a freezer until bag contents were analyzed. Descriptions, illustrations, and keys in Miller (1961), Wagner (1978), Akre *et al.* (1980), and Eck (1984) were used to identify captured vespine wasps. Taxonomy used here follows that of Carpenter and Kojima (1997). Voucher specimens are deposited in the James Entomological Collection at Washington State University, Pullman, WA, and with the USDA, ARS Subarctic Agricultural Experiment Station in Fairbanks, AK.

For each species, means for wasps captured per trap per week in 2003 were compared between chemical attractant treatments using ANOVA and Tukey's test (DataMost 1995) to determine differences among means. Similar data for 2004 were compared using a paired *t*-test. For developing seasonality profiles for each species, numbers of wasps captured in each trap per week were averaged for the traps at each site. Unless stated otherwise, data analyses and results are for worker wasps.

RESULTS

The most abundantly trapped wasp in both years and at all three study locations was *V. vulgaris*. In 2003, 508 worker *V. vulgaris* were captured in traps. In 2004, 4 queens, 1825 workers, and 36 males of *V. vulgaris* were captured. In 2003, *V. vulgaris* workers were primarily in traps baited with the combination of acetic acid and isobutanol, with no or few wasps in unbaited traps and traps baited with acetic acid, isobutanol, heptyl butyrate, or acetic acid plus heptyl butyrate (Table 1). In 2004, *V. vulgaris* workers again were primarily in traps baited with acetic acid plus isobutanol, with nearly none in traps baited with heptyl butyrate (Table 2). The same pattern was seen for *V. vulgaris* males in traps (Table 2). The four *V. vulgaris* queens were captured in traps (in late May and early June) baited with acetic acid plus isobutanol. Workers were captured between mid June and early October, and males from late July into late September. Workers were most abundantly trapped from mid July into late August 2004 (Figure 1A).

In 2003, 9 worker *V. acadica* were captured in traps, primarily in traps baited with heptyl butyrate (Table 1). In 2004, the 42 worker and 4 queen *V. acadica* captured were all in traps baited with heptyl butyrate

Table 1.

Mean ± SE numbers of wasps captured per trap, for unbaited traps (CONTROL), and for traps baited with acetic acid (AA), isobutanol (IB), heptyl butyrate (HB), acetic acid plus isobutanol (AAIB), and acetic acid plus heptyl butyrate (AAHB). Fairbanks, Alaska, 2003.

Wasps (workers)[1]	CONTROL	AA	IB	AAIB	HB	AAHB
V. vulgaris	5.0 ± 1.8a	8.0 ± 4.3a	11.2 ± 5.6a	60.0 ± 32.9b	1.6 ± 1.4a	15.8 ± 10.0a
D. maculata	0.0 ± 0.0a	12.2 ± 7.1c	1.6 ± 0.7ab	7.6 ± 3.1bc	0.6 ± 0.6a	4.8 ± 2.3ab
V. acadica	0.0 ± 0.0a	0.0 ± 0.0a	0.0 ± 0.0a	0.0 ± 0.0a	1.6 ± 0.8b	0.2 ± 0.2a

[1] For each species, means followed by the same letter are not significantly different at $P \leq 0.05$ by Tukey's test. N = 5.

Table 2.
Mean ± SE numbers of wasps captured per trap, for traps baited with heptyl butyrate (HB) and for traps baited with acetic acid plus isobutanol (AAIB). Fairbanks, Delta Junction and Palmer, Alaska, 2004.

Wasps[1]	HB	AA/IB
V. vulgaris workers	0.46 ± 0.27a	140.18 ± 46.10b
V. vulgaris males	0.07 ± 0.07a	3.08 ± 0.87b
D. maculata workers	0.31 ± 0.17a	33.92 ± 15.17b
D. maculata queens	0.00 ± 0.00a	2.0 ± 0.72b
V. acadica workers	3.54 ± 0.91b	0.00 ± 0.00a
V. consobrina workers	1.38 ± 0.53b	0.00 ± 0.00a
V. rufa workers	1.54 ± 0.42b	0.00 ± 0.00a

[1] For each species, means followed by the same letter are not significantly different at $P \leq 0.05$ by a paired *t*-test. N = 13.

(Table 2). Three of the queens were captured in late June and one in mid September. Workers were captured from mid June to mid September 2004 (Figure 1B).

In 2003, only three *V. consobrina* and one *V. rufa* were captured; all were workers in traps baited with heptyl butyrate. In 2004, 17 *V. consobrina* and 20 *V. rufa* workers were captured, all in traps baited with heptyl butyrate (Table 2). *Vespula consobrina* workers were captured in traps from 7 July to 3 August in Delta Junction and from 12 June to 19 July in Fairbanks. Worker *V. rufa* were captured in traps from 29 June to 27 July in Delta Junction, from 12 June to 19 July in Fairbanks, and two were in traps on 5 August in Palmer.

In 2003, 134 worker *D. maculata* were captured. Numbers of bald-faced hornets in traps baited with acetic acid, and with acetic acid plus isobutanol, were significantly greater than in unbaited traps (Table 1). In 2004, 445 worker and 27 queen *D. maculata* were captured. Most were captured in traps baited with acetic acid plus isobutanol. In both years, very few were captured in traps baited with heptyl butyrate (Table 2). No male *D. maculata* were captured in these traps. Queens were captured from mid May to early June, and workers from late June into late August (Figure 1C).

In this study, too few *D. arenaria* (9) *D. norvegicoides* (2), or *D. norwegica* (2) wasps were captured for any statistical analysis, while no *D. alpicola*, *D. adulterina*, or *V. austriaca* wasps were captured.

DISCUSSION

The species captured in traps during this study in the vicinities of Fairbanks, Delta Junction, and Palmer, Alaska, vary somewhat from those species reported by Miller (1961), Akre *et al.* (1980), Eck (1984) and Carpenter and Kojima (1997). *Vespula consobrina* were captured in traps at two of the three sites (Fairbanks and Delta Junction), despite its indicated absence from most of Alaska by both Miller (1961) and Akre *et al.* (1980). The nearest collection locations in those references are in the southernmost Alaska panhandle, northern British Columbia, and southeastern Yukon. The absence of *D. adulterina*, *D. alpicola*, and *V. austriaca* in traps could have been the result of their absence in the areas trapped, or a lack of response to the lures.

The seasonal patterns of wasp captures in traps indicate a broad period during which they could be pestiferous; from early July to early September. Of most interest as a pest is the common wasp *V. vulgaris*, because of its abundance and its scavenging

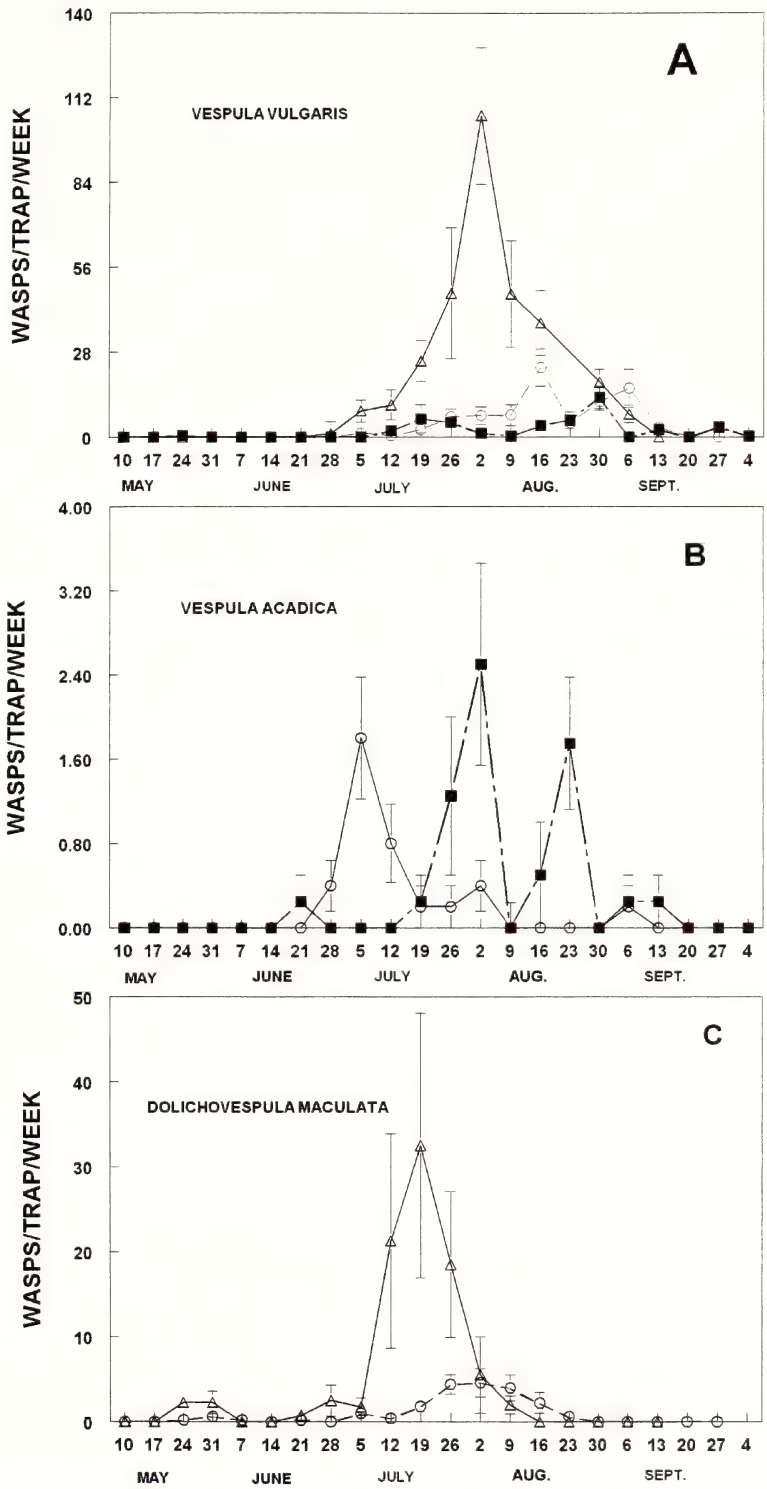

Figure 1. Mean ± SE numbers of female *V. vulgaris* (A), *V. acadica* (B) and *D . maculata* (C) in traps baited with acetic acid and isobutanol, through the 2004 field season. Lines do not imply dependence of data. Sites are Fairbanks (open triangles), Delta Junction (filled squares), and Palmer (open circles).

behavior which bring it into frequent contact with people (Akre *et al.* 1980). The bald-faced hornet, *D. maculata,* may also be pestiferous in Alaska due to its abundance during July and August. Other species of wasps captured, such as *V. acadica,* and *V. consobrina*, are less likely to be pestiferous because they are not known for scavenging habits, occur in smaller colonies, and do not occur in high densities, compared to species such as *V. vulgaris* (Akre *et al.* 1980).

The patterns of responses of wasps in Alaska to different chemical attractants are consistent with results from trapping studies in other areas of North America. The three members of the *V. rufa* species group (*V. acadica*, *V. consobrina*, and *V. rufa*) were attracted to heptyl butyrate, and were not captured in significant numbers in traps baited with acetic acid plus isobutanol. This pattern was seen in Washington State, where *V. atropilosa*, a *V. rufa* species group member, was attracted to heptyl butyrate and not to acetic acid plus isobutanol (Landolt 1998), and in Michigan, where *V. consobrina* and *V. vidua*, both *V. rufa* species group members, exhibited the same response pattern (Reed and Landolt 2002). In the present study, the only member of the *V. vulgaris* species group present was *V. vulgaris.* Unlike species in the *V. rufa* group, it was attracted to acetic acid plus isobutanol and not to heptyl butyrate. This pattern matches results of earlier studies (Landolt 1998, Landolt *et al.* 1999, Reed and Landolt 2002), where *V. flavopilosa* Jacobson, *V. germanica*, *V. maculifrons*, and *V. vulgaris,* all *V. vulgaris* species group members, were trapped with acetic acid plus isobutanol and not with heptyl butyrate. *Vespula pensylvanica* (another *V. vulgaris* group member), however, is clearly attracted to both lures (Landolt 1998). The response by *D. maculata* to acetic acid plus isobutanol and lack of a response to heptyl butyrate is consistent with earlier studies. In Maryland and in western Washington (Landolt *et al.* 2001), as well as in Michigan (Reed and Landolt 2003), *D. maculata* workers were trapped with acetic acid, but more so to acetic acid plus isobutanol. In this study, numbers of workers of *D. maculata* captured in traps baited with acetic acid plus isobutanol were not significantly higher than with acetic acid alone. The small numbers of workers of *D. arenaria*, *D norvegicoides*, and *D. norwegica* trapped here are not suitable for statistical analyses, and indicate either a very weak response to the lures, or very low population densities.

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Redescription of *Haliplus dorsomaculatus* (Coleoptera: Haliplidae) with a New Synonymy and Comments on Habitat and Distribution

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ABSTRACT

Adults of *Haliplus dorsomaculatus* Zimmermann are redescribed including a discussion of male and female morphological characters. The species ranges from southern British Columbia east to western Montana and south to northeastern Utah and northern California; there is little geographic variation. Its preferred habitat appears to be emergent vegetation near the margins of slowly flowing water. *Haliplus allisonae* Brigham is a junior synonym of *H. dorsomaculatus*.

Key Words: *Haliplus allisonae*, Nearctic, wing venation, mandible, female genital sclerites

INTRODUCTION

Haliplidae is a small family of aquatic adephagan beetles most obviously characterized by the greatly expanded metacoxal plates. There are about 65 species in North America; these are placed in four genera, the largest of which is *Haliplus* Latreille. Wallis (1933) revised the Nearctic *Haliplus* species; six species have subsequently been described (Mank 1940; Leech 1948; Brigham & Sanderson 1972, 1973; Brigham 1977; Wells 1989). The Nearctic members of *Haliplus* have been assigned to three subgenera, *Haliplus sensu stricto*, *Liaphlus* Guignot and *Paraliaphlus* Wallis.

Haliplus dorsomaculatus Zimmermann (1924) was described from a single male specimen from "Boreal America". It belongs to the nominotypical subgenus (Guignot 1928, see discussion in Holmen 1987, p. 90), characterized by the presence of pronotal plicae and penultimate segments of the labial palps produced on the medioapical angle. Wallis (1933) did not examine the type specimen for this species and uncertainly referred to a male from CA and a female from CO as "? *dorsomaculatus*". Based on these specimens and his interpretation of Zimmermann's description, Wallis provided a new description and included *H. dorsomaculatus* in his key to species. Certain characters in his key do not apply to the *H. dorsomaculatus* holotype and his illustration of the male genitalia is incorrect for that species. These errors have long been suspected; a specimen correctly identified by H.B. Leech in 1946 has a note attached: "Can this be the true *H. dorsomaculatus* Zimm?"

More recently, Brigham (1977) described a new species, *Haliplus allisonae*, based on a series of specimens from BC similar to those he previously correctly identified as *H. dorsomaculatus*. Brigham discussed separating his new species from *H. distinctus* Wallis but mentioned *H. dorsomaculatus* only in his amended version of Wallis's key. Here it is shown that *H. allisonae* is a junior synonym of *H. dorsomaculatus*.

Apart from Zimmermann's brief description, little has been published about *H. dorsomaculatus*. No habitat information is available and, due to the high level of misidentifications, all published distribution information should be considered suspect. Here, the habitat preference for *H. dor-*

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somaculatus is described and its currently known distribution, based on examined specimens, is mapped.

In water beetles, as with most organisms with male intromittent organs (Eberhard 1985), male genitalia frequently provide unequivocal characters for the identification of species. Female characters are often overlooked and in many cases only males can be identified with certainty. Galewski (1972a, 1972b) showed that European haliplid females can be identified by mandibular and external genital characters and more recent guides to these species have included illustrations of the latter (Franciscolo 1979, Holmen 1987). Here, these characters and the metathoracic wing are illustrated for *H. dorsomaculatus* and a brief description of the internal structures of the female reproductive system is given.

MATERIALS AND METHODS

In addition to the type specimens of *H. dorsomaculatus* and *H. allisonae*, 718 *H. dorsomaculatus* from the museums and private collections listed in Table 1 were examined. Collection information for all *H. dorsomaculatus* specimens examined is listed in the text (type specimens) or Appendix 1. Standard postal abbreviations for states and provinces are used throughout.

Using a calibrated ocular micrometer on a Wild M5 stereomicroscope with 10x eyepieces, the following measurements were taken: L, distance from the front of the head to the apices of the elytra in lateral view; W, maximum width across the elytra in dorsal view; IO, minimum distance between the eyes in dorsal view; and H, maximum width of the head measured across the eyes in dorsal view. Five to eight specimens of each sex were measured for a given locality; means and standard errors are reported. Where insufficient numbers of specimens from a single locality were available, those from localities in the same county were combined. The normalized interocular distance, Rel. IO, was calculated using Rel. IO = IO / H.

Many male specimens were dissected in order to examine the genitalia. For freshly collected specimens, the genitalia were simply extended, spread and allowed to dry in place. Older specimens were relaxed in hot water and the genital capsule removed using fine forceps. The capsule was cleared of non-sclerotized tissue in hot 10% aqueous KOH and the genitalia were teased out of the capsule. After examination, the genitalia and capsule were stored in glycerin in genitalia vials on the same pin as the specimen. Some females were dissected in a similar manner to characterize the external genitalia. Other females were dissected following a procedure similar to Mazzoldi (1996) and Miller (2001a), using 0.2% aqueous Toluidine Blue to stain the preparation before microscopic examination. Mandibles or metathoracic wings were removed from several specimens and examined in temporary mounts on microscope slides. Nomenclature for wing venation follows Ward (1979). Drawings were made using a drawing tube on a Wild M5 stereomicroscope.

RESULTS

Haliplus dorsomaculatus Zimmermann 1924.

Holotype: ♂, "Amer. bor.", no date, no collector, ZSMC.

Haliplus allisonae Brigham 1977. Holotype: ♂, CANADA, BC, Creston, King Cr., 22 Sept 1955, G. Stace-Smith, INHS; Allotype: ♀, same data, INHS; Paratypes: 4 ♂, 4 ♀, same data; INHS; 1 ♂, 1 ♀, same data, CAS. NEW SYNONYMY.

For habitus drawings, see Brigham (1977, Fig. 1) and Hatch (1953, Plate XXXIII Fig. 3 - note the figure is incorrectly labeled as *H. longulus* LeConte and is broader (L/W = 1.75), has a larger Rel. IO (=0.63) and shorter pronotal plicae than

Table 1.
Sources of Specimens examined in this study with abbreviations and contact person.

Abbr.	Source	Contact
AMNH	American Museum of Natural History, New York, New York	L. H. Herman
BCPM	Royal British Columbia Museum, Victoria, BC	R. A. Cannings
BYU	Monte L. Bean Life Science Museum, Brigham Young University, Provo, UT	R. W. Baumann
CAS	California Academy of Sciences, San Francisco, CA	D.H. Kavanaugh
CBBC	Cheryl Barr Collection, c/o Essig Museum, University of California, Berkeley, CA	C. B. Barr
CNC	Canadian National Collection of Insects, Agriculture and Agri-Food Canada, Ottawa, Ontario	Y. Bousquet
EMEC	Essig Museum of Entomology, University of California, Berkeley, CA	C. B. Barr
INHS	Illinois Natural History Survey, Champaign, Illinois	C. Favret
JBWM	J.B. Wallis Museum, University of Manitoba, Winnipeg, Manitoba	R. E. Roughley
MTEC	Montana Entomological Collection, Montana State University, Bozeman, MT	M. A. Ivie
NSNH	Nova Scotia Museum of Natural History, Halifax, Nova Scotia	C. Majka
OSAC	Oregon State Arthropod Collection, Oregon State University, Corvallis, OR	D. Judd
RDKC	Rex D. Kenner Collection, Vancouver, BC	R.D. Kenner
ROME	Royal Ontario Museum, Toronto, Ontario	D. Currie
UASM	Strickland Museum, University of Alberta, Edmonton, AB	D. Shpeley
UBCZ	Spencer Entomological Museum, University of British Columbia, Vancouver, BC	K. M. Needham
WSU	Maurice T. James Entomological Collection, Washington State University, Pullman, WA	R. S. Zack
ZSMC	Zoologische Staatssammlung München, München, Germany	M. Baehr

normal for *H. dorsomaculatus*).

Holotype. Male on point with labels: white rectangular "Amer. bor."; circular white "Typop" handwritten; red rectangular "Typus" printed; white rectangular "Samml. A. Zimmermann"; red rectangular "Holotype ♂ Haliplus dorsomaculatus Zimmermann 1924", species handwritten; blue rectangular "Zool. Staatsslg. München". Genitalia on point on separate pin with labels: white rectangular "penis H. dorsomaculatus" handwritten; white rectangular "Samml. A. Zimmermann"; red rectangular "Holotype Haliplus dorsomaculatus Zimmermann 1924", species handwritten; blue rectangular "Zool. Staatsslg. München".

L = 3.32 mm, W = 1.62 mm, L/W = 2.05, Rel. IO = 0.51. Elongate oval, fairly pointed posteriorly; not very convex, maximum near midpoint. Head amber with darker post-ocular band extending to middle of compound eyes; labrum medially emarginate with dense fringe of setae, medial dark mark extending to clypeus, micropunctation sparser medially and anteriorly; clypeus coarsely punctured, narrow impunctate area extending onto frons; interocular area coarsely punctured with inverted U-shaped impunctate area; dense punctures along ocular margin; palpi same colour as head, not infuscate apically, labial palpi with penultimate segment produced medially; vertical carina behind eye on side of

head. Pronotum yellow, paler than head; narrow medial impunctate area extending approximately 2/3 along midline from posterior margin, ovate impunctate area medially each side of the midline, puncturing less dense anterior to these impunctate areas; plicae deeply impressed with brown lateral edges, impression steep on lateral side, sloping more gradually medially; plicae approximately 1/3 length of pronotum measured along plical line; pronotal lateral bead ends just anterior to posterolateral corner; pronotum wider than elytral base by width of lateral bead; hind margin sinuate with point of inflection slightly medial to midpoint of each side; anterior part of hypomeron visible in lateral view; lateral margin of pronotum evenly curved ventrally from posterior to anterior; anterolateral corner about midpoint of eye. Elytra same colour as pronotum with brown maculation as follows: sutural blotch extending to stria 3 anteriorly, stria 4 posteriorly; medial discal blotch between striae 3 and 4 with posteromedial corner connected to sutural blotch; postmedial discal blotch between striae 5 and 7; weak indications of premedial discal blotch between striae 5 and 6, most obvious along stria 6 where 4 or 5 punctures have merging "brown halos". Punctures of striae 1-4 large and blackened, basal punctures of striae 2 and/or 3 enlarged; punctures of striae 5-10 decreasing in size laterally, those of stria 9 similar in size to interstrial punctures, stria 10 with even smaller, barely blackened punctures. Sutural interstrial row with punctures nearly linear and single with occasional doubled or misplaced punctures; subsequent interstrial rows with punctures more widely spread, especially apically where some rows obsolete; no micropunctures visible at 50x between larger punctures; elytral apical margin very weakly sinuate. Prosternal process with sides converging to minimum of constriction, which occurs at about anterior margin of procoxae, subsequently widening posteriorly to just short of apex, then nearly parallel to apex; not channeled anteriorly, very shallowly channeled from anterior end of constriction to apex, flat between margins; coarsely punctured with dense micropuncturing from near bottom of declivity to posterior margin;. Metasternum mostly impunctate, a few larger punctures laterally; in ventral view, weakly depressed behind mesocoxae. Ventral surface similar in colour to dorsal surface, legs somewhat redder; micropuncturing as described in Brigham (1977). Genitalia as in Brigham (1977, Figs. 2–4) except aedeagus with distal end of "dorsal hump" somewhat farther from apex (fragments of tissue on dorsal edge suggest "hump" originally in exact agreement). Protarsi only slightly produced, with specialized setae; protarsal claws about equal in length but anterior claw more sharply bent near base and somewhat broader and thicker than posterior claw. Mesotarsi slightly produced, with specialized setae; mesotarsal claws equal, longer and more gently curved than protarsal claws.

Males. L = 3.16 ± 0.02 mm, W = 1.65 ± 0.01 mm, L/W = 1.92 ± 0.01, Rel. IO = 0.53 ± 0.002 (n=50; no significant geographical variation observed in these characters, see Table 2); as in holotype except as follows: medial discal blotch usually merged with sutural blotch to give triangular sutural blotch with apex pointed posteriorly; one to three lateral blotches on each elytron (no geographic pattern to variation in maculation); in a minority of specimens, apex of aedeagus slightly more tapered to form a narrower tip (no geographic pattern except tapered condition more prevalent in CA specimens). Mandibles and metathoracic wing as in females (see below).

Females. L = 3.13 ± 0.02 mm, W = 1.65 ± 0.01 mm, L/W = 1.89 ± 0.01, Rel. IO = 0.54 ± 0.003 (n=49; no significant geographical variation observed in these characters, see Table 2); similar to males except as follows: protarsal claws slender, evenly curved, equal; pro- and mesotarsi not produced and without specialized setae. External genital sclerites, mandibles and metathoracic wing as shown in Figs. 1–3. Internal structures of female reproductive system weakly sclerotized, spermathecal duct short, such that spermatheca ventral to

Table 2.
Length, width and relative interocular distance for *H. dorsomaculatus* as a function of geographic location.

Locality	L(mm) males	W(mm) males	L/W males	Rel. IO males	L(mm) females	W(mm) females	L/W females	Rel. IO females
CA, Lassen Co.	3.21 ± 0.05	1.62 ± 0.03	1.98 ± 0.01	0.53 ± 0.004	3.17 ± 0.05	1.66 ± 0.03	1.91 ± 0.01	0.56 ± 0.007
OR, Multnomah Co.	3.25 ± 0.06	1.74 ± 0.02	1.87 ± 0.04	0.51 ± 0.007	3.27 ± 0.03	1.75 ± 0.02	1.87 ± 0.03	0.53 ± 0.007
WA, Pierce Co.	3.09 ± 0.02	1.61 ± 0.02	1.92 ± 0.02	0.53 ± 0.002	2.98 ± 0.06	1.55 ± 0.03	1.92 ± 0.01	0.53 ± 0.006
WA, King Co.	3.06 ± 0.05	1.56 ± 0.03	1.96 ± 0.01	0.53 ± 0.009	3.07 ± 0.03	1.62 ± 0.01	1.90 ± 0.02	0.53 ± 0.005
BC, Surrey	3.11 ± 0.03	1.62 ± 0.02	1.92 ± 0.02	0.51 ± 0.006	3.10 ± 0.02	1.64 ± 0.01	1.89 ± 0.01	0.54 ± 0.006
BC, Wynndel	3.27 ± 0.05	1.68 ± 0.01	1.95 ± 0.02	0.54 ± 0.004	3.17 ± 0.05	1.67 ± 0.03	1.90 ± 0.01	0.55 ± 0.010
ID, Clearwater Co.	3.12 ± 0.05	1.75 ± 0.03	1.79 ± 0.02	0.53 ± 0.008	3.10 ± 0.03	1.71 ± 0.01	1.81 ± 0.02	0.55 ± 0.006
MT, Gallatin Co.	3.16 ± 0.03	1.63 ± 0.01	1.94 ± 0.01	0.54 ± 0.005	3.13 ± 0.04	1.63 ± 0.02	1.92 ± 0.01	0.56 ± 0.005
UT, Cache Co.	3.15 ± 0.04	1.67 ± 0.02	1.89 ± 0.02	0.52 ± 0.008	3.19 ± 0.02	1.68 ± 0.01	1.90 ± 0.005	0.53 ± 0.006
All specimens	3.16 ± 0.02	1.65 ± 0.01	1.92 ± 0.01	0.53 ± 0.002	3.13 ± 0.02	1.65 ± 0.01	1.89 ± 0.01	0.54 ± 0.003

apex of vagina; bursa copulatrix connected ventrally to vagina between spermatheca and apex of vagina.

Diagnosis. Among the Nearctic members of *Haliplus s. str.*, the adults of *H. dorsomaculatus* are most similar to those of *H. longulus*; these two species share a similar elongate shape and size. They may most easily be separated by the characters of the prosternal process. In *H. dorsomaculatus*, the prosternal process is essentially flat in cross-section between the margins with dense micropuncturing between the abundant larger punctures. In *H. longulus*, the cross-section is convex with sparse coarse punctures and no micropunctures.

Other external characters that help to separate the adults of these species are the pronotal plicae and the elytral maculation. In *H. dorsomaculatus*, each plica is asym-

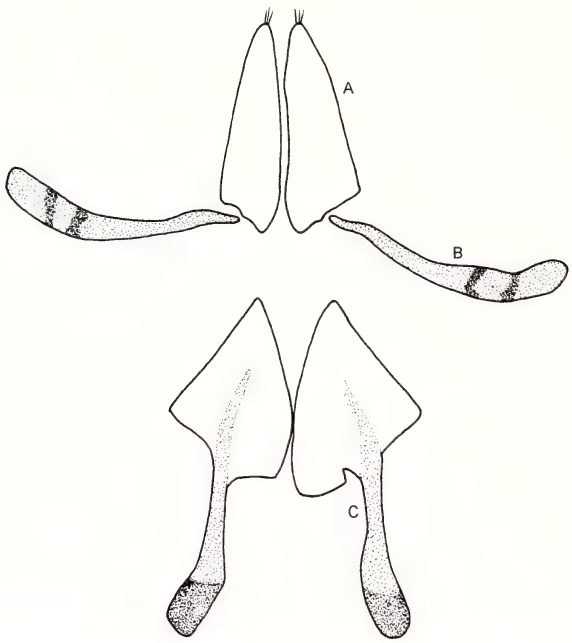

Figure 1. Female genital sclerites of *Haliplus dorsomaculatus*, posterior up; (A) gonocoxae, (B) tergal halves IX, (C) gonocoxosternites. USA, ID, Benewah Co., E Fork of Charles Creek, 27 July 1987, R.S. Zack, WSU.

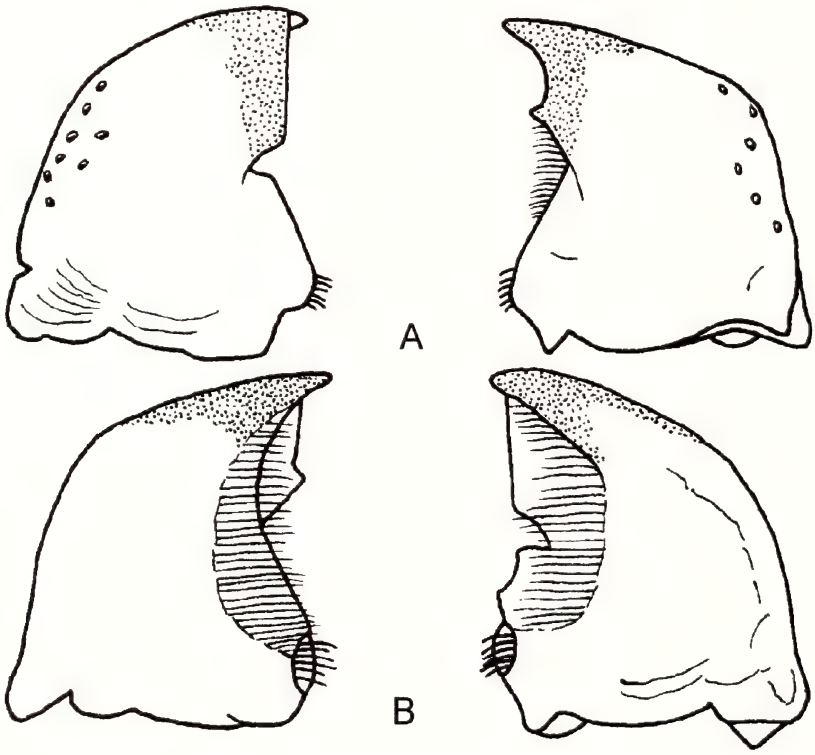

Figure 2. Mandibles of *Haliplus dorsomaculatus*, female, anterior up; (A) dorsal view, (B) ventral view. Canada, BC, Wynndel, Head of Lizard Creek, 07 April 1946, G. Stace-Smith, SEM #3142.

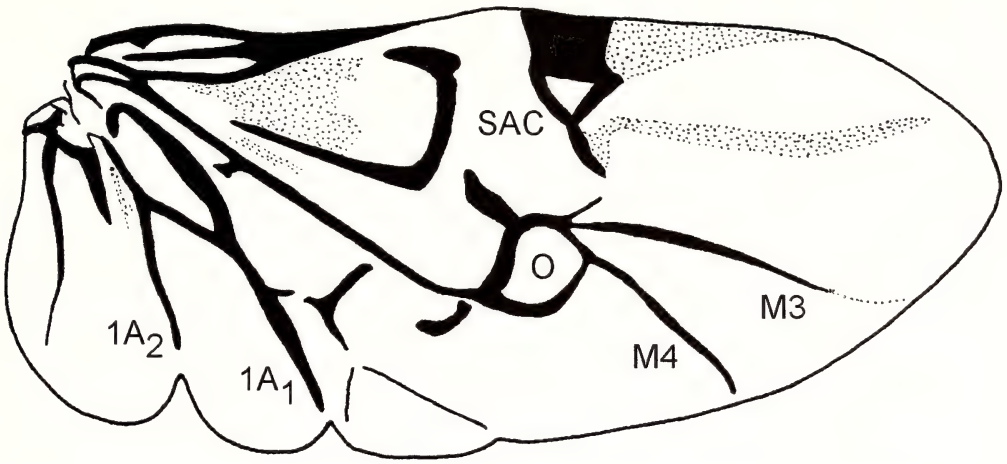

Figure 3. Right metathoracic wing of *Haliplus dorsomaculatus*, ventral view, female, O: oblongum cell; SAC: anterior sector cell, M: medial veins, A: anal veins, nomenclature according to Ward (1979). Canada, BC, Surrey, 110 Ave. right-of-way N of 168 St., 03 September 2004, R.D. Kenner, RDKC.

metrically impressed, usually with an infuscate lateral margin. In *H. longulus*, each pronotal plica is symmetrically impressed and is not infuscate. In *H. dorsomaculatus*, the sutural blotch is roughly triangular with the apex pointed posteriorly and there are one to three lateral blotches on each elytron. In *H. longulus*, the sutural blotch is not roughly triangular and may be almost obsolete; there are zero to one lateral blotches on each elytron.

Males of *H. dorsomaculatus* and *H. longulus* can also be separated by differences in their aedeagi. In *H. dorsomaculatus*, the aedeagus is shaped like "an inverted boot" (Zimmermann 1924), see Brigham (1977, Fig. 3). In *H. longulus*, the main axis of the aedeagus is gently curved and the apical quarter is narrowed, see Wallis (1933, Fig. 9d), Hilsenhoff & Brigham (1978, Fig. 5T), Gundersen & Otremba (1988, Fig. 66), Durfee, Jasper & Kondratieff (2005, Fig. 3).

Distribution. *Haliplus dorsomaculatus* ranges along the coastal mountains from northern CA to southern BC, east to western Montana and south along the Rocky Mountains to northeastern UT (Fig. 4). No records are known from south or east of the Great Divide Basin in WY. This may be an artifact as relatively little material from either WY or CO was available for this study. Durfee, Jasper & Kondratieff (2005) list *H. dorsomaculatus* as "unconfirmed" in CO. All examined specimens from CO and CA previously determined as *H. dorsomaculatus* were misidentified. A long series of *H. dorsomaculatus* from Lassen Co., CA was found in the unidentified material from the CAS. This series is the only valid record for CA known to the author.

Habitat. Many of the collection locations for *H. dorsomaculatus* imply lotic habitats although some specimens appear to have been collected in lentic habitats. Descriptions of habitats at some collection sites have been provided by R. S. Zack (pers. comm.), and R. W. Baumann (pers. comm.). Zack reported taking haliplids by digging or kicking into emergent vegetation along the margins of generally slowly flowing water. Baumann reported that all of his sites were associated with spring-fed creeks.

In 2004, *H. dorsomaculatus* was collected from three locations in the Lower Fraser Valley, BC: an apparently permanently flooded drainage ditch beside a highway, a very shallow, apparently spring-fed pool beside an industrial parking lot and a small creek draining an apparently spring-fed swamp at the base of bluffs on the edge of the Fraser River flood plain. Only two and three specimens, respectively, were

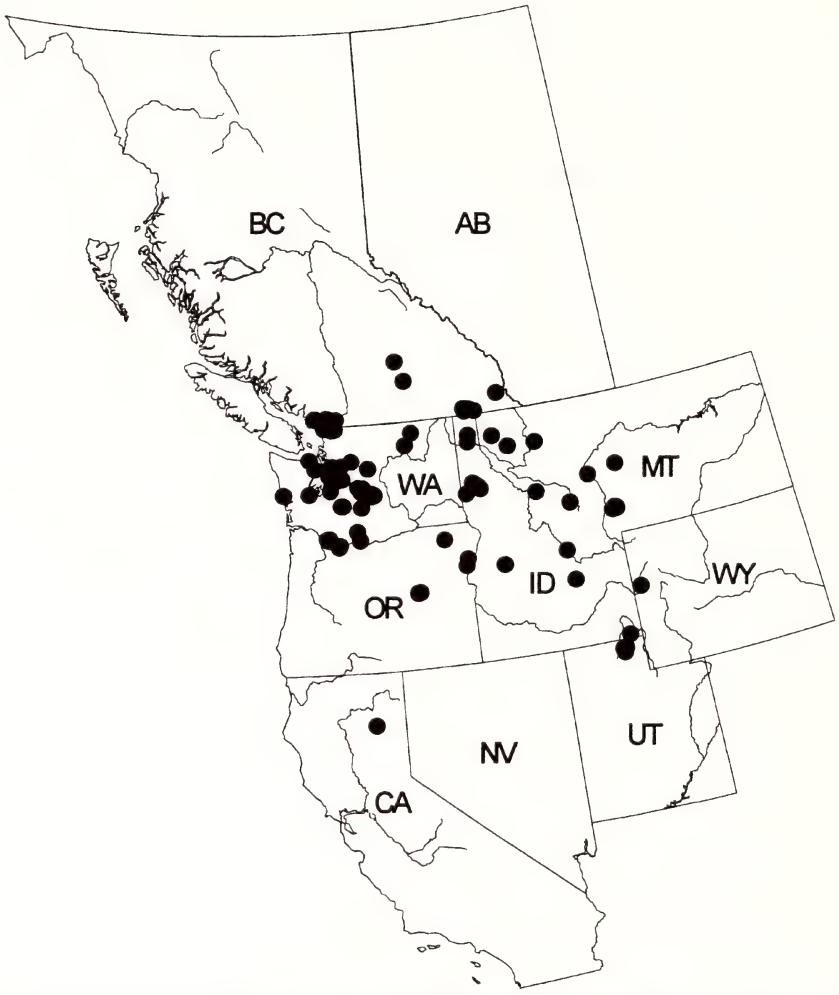

Figure 4. Distribution of *Haliplus dorsomaculatus* based on specimens examined by the author. Collection data given in Appendix 1.

collected from the first two sites. The creek site yielded a good series, including probable teneral specimens, all from dense emergent vegetation near the creek margin; no *H. dorsomaculatus* were found in other microhabitats in the creek.

DISCUSSION

The confusion surrounding the identification of *H. dorsomaculatus* stems chiefly from errors in Wallis (1933). It is likely that Wallis had no *H. dorsomaculatus* specimens in the material he examined. The apparent CA specimen is possibly *H. robertsi* Zimmermann although the species limits and status of that taxon are not clear. The CO specimen has not been found.

Two couplets in Wallis's key can give problems in determining *H. dorsomaculatus*: those concerning: (i) channeling of the prosternal process and (ii) the characters of the mid-metasternum and male protarsal claws. The first couplet requires a subjective choice between "evidently" and "very feebly or not" channeled. The prosternal process of *H. dorsomaculatus* can easily be described as "very feebly channeled". That choice leads to *H. longulus* and many previous determinations have reached that conclusion. The second couplet requires a mid-metasternum with lateral longitudinal impressions and male protarsal claws equal.

The mid-metasternum of *H. dorsomaculatus* has coarse punctures laterally and, in some specimens, these overlap to produce what could be described as a longitudinal impression. However, the male protarsal claws are never equal. The other half of this couplet leads to *H. distinctus* Wallis. See Brigham (1977) for a discussion of the differences between *H. dorsomaculatus* (as *H. allisonae*) and *H. distinctus*.

Comparison of the holotypes of *H. dorsomaculatus* and *H. allisonae* makes it clear that these are the same species. The only significant difference between the two specimens is in the base colour: yellow for *H. dorsomaculatus* and reddish brown for *H. allisonae*. The colour of the *H. allisonae* type series is likely an artifact of specimen preparation or storage because other specimens collected by Stace-Smith at the *H. allisonae* type locality over a two-week period (which includes the collection of the type series) are the "normal" yellow colour and similar darker specimens occasionally occur in preserved specimens of other species.

Female reproductive characters. Galewski (1972a) showed that, although interspecific differences among the characters of the external female genitalia can be subtle, there are larger differences at higher taxonomic levels. Drawings of these sclerites showing greater detail have subsequently been published for the European species (Franciscolo 1979; Holmen 1987). The external genitalia of *H. dorsomaculatus* (Fig. 1) are, most similar to those of *H. wehnckei* Gerhardt (=*H. sibiricus* Motschulsky, see Lundmark, Drotz & Nilsson (2001)) illustrated in Holmen (1987, Fig. 251). Characters of the gonocoxae should be used with caution, since these structures tend to collapse or distort upon drying and are difficult to characterize in drawings. Further study of the external female genital sclerites will need to be done before their diagnostic utility for the North American species can be determined.

Characters of the internal female reproductive system, especially the spermatheca, have proven useful both in the determination of phylogenetic relationships and in species determinations (Burmeister 1976, Ordish 1985, Mazzoldi 1996, Miller 2001a, 2001b). It is anticipated that these characters may be similarly useful in haliplids (Holmen 1987). Burmeister (1976) investigated the internal parts of the female reproductive system in *Haliplus (Neohaliplus) lineatocollis* (Marsham) although he did not investigate the spermatheca in detail. His findings differ from what was observed here for *H. dorsomaculatus*; they are, however quite similar to what was found for *Haliplus (Liaphlus) gracilis* Roberts (RDK, unpublished data). In *H. lineatocollis*, the spermathecal duct is relatively long and the bursa copulatrix is connected to the anterior end of the vagina (Burmeister 1976, Fig. 44b) whereas in *H. dorsomaculatus*, the spermathecal duct is short and the bursa copulatrix is connected ventrally to the vagina. With no other species for comparison, it is hard to draw conclusions at this point but these results do suggest that these characters deserve further investigation.

Mandibular and metathoracic wing characters. Galewski (1972b) suggested the use of mandibular characters for the identification of haliplid females. However, in this work no difference in mandibular characters was found between conspecific males and females suggesting that these characters may also be useful for the identification of males. Although the differences in these characters between species can be subtle, they are potentially useful at the species and higher taxonomic levels (Galewski 1972b, RDK, unpublished data). Within the *Haliplus s. str.* species shown by Galewski, the relative size of the apical tooth of the right mandible appears to be a useful character. Comparing the mandibles of *H. dorsomaculatus* (Fig. 2) with those shown in Galewski (1972b, Figs. 1–5, 7–10), *H. dorsomaculatus* is, as above, most similar to *H. wehnckei*. Until information is available for more Nearctic species, it is hard to determine how useful mandibular characters will be for species identification in North America.

Characters of the metathoracic wings

are little used in the identification of beetles, in part, because venation shows little variation between species in many groups of beetles. Ward (1979) discussed three wing venation characters that appeared to be useful in adephagan beetles: (i) the shape of the oblongum cell, (ii) the position of the distal segment of M_4 relative to M_3 and Cu_1 and (iii) the position of the SA vein which separates the SA cell from the 3R cell.

There appear to be few published examples of haliplid metathoracic wing venation comparable to that shown in Fig. 3. Balfour-Browne (1943) examined 13 species of Haliplidae but illustrated only the area around the oblongum cell for *Peltodytes caesus* (Duftschmidt) (Balfour-Browne 1943, Fig. 18). He discussed general trends in the venational characters of adephagan beetles, including haliplids, but it is difficult to make detailed comparisons in the absence of drawings. Illustrations of the wings of three haliplid species have been published: *Haliplus (Haliplus) ruficollis* De Geer (Franciscolo 1979), *Peltodytes muticus* (LeConte) (Wallace & Fox 1980) and *Haliplus (Liaphlus) fulvus* (Fabricius) (Holmen 1987). In addition, a Master's thesis (Mousseau 2004) includes drawings for the three Nearctic *Brychius* species.

Even within a single genus, the overall shape of the metathoracic wing can vary (Mousseau 2004). In the *Haliplus* figures referenced above, the posterior margin of the wing is smooth and continuous, or nearly so. In *H. dorsomaculatus* the posterior margin is relatively deeply emarginate at the position of the anal fold (approximately aligned with the distal end of the $1A_2$ vein) and, to a lesser extent, at the end of the $1A_1$ vein (Fig. 3).

The wing venation is identifiably different in the figures for each of the seven species listed above. The three *Haliplus* species differ in the shape of the oblongum cell and the relative position of the distal portions of M_3 and M_4. The position of the SA vein appears to be similar in all three species. These results suggest that in Haliplidae, wing venation may provide diagnostic characters at the species level, although more work is needed to determine the limits of intraspecific variation.

Distribution and habitat. *Haliplus dorsomaculatus* appears to be a strictly western Nearctic species (Fig. 4). A record from NF (Larson 1987, Roughley 1991) is almost certainly an error. No voucher specimen has been found to support that record and the original source is unknown. *Haliplus dorsomaculatus* should be removed from the NF list until a verifiable voucher specimen has been found.

In western North America, the distribution for *H. dorsomaculatus* corresponds to the mountainous areas. This correlation is particularly apparent in WA where, in eight decades of collecting, there are there are records from the Olympic Mountains and almost every county overlapping the Cascade Mountains but none from eastern WA . *Haliplus dorsomaculatus* may eventually be found in extreme southeastern WA as there are records from the neighbouring Blue Mountains in northeastern OR. The correlation with mountainous terrain fits well with a preferred habitat associated with relatively permanent flowing water, as springs or creeks need sources of water at higher elevations.

Further collecting is needed to clarify the distribution in several areas. Given the known distribution in southern BC and western MT, it seems likely that *H. dorsomaculatus* will be collected in southwestern AB. More collecting is needed in OR to determine if the apparent gap in the distribution between northern CA and northern OR is real. The southern limits, both in CA and the Rocky Mountains, need further investigation. In UT, *H. dorsomaculatus* appears limited to the northeastern corner; its status in the Uinta Mountains is uncertain. With the habitat information given here, it should be possible to conduct directed searches to clarify the status of this species in all of these areas.

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APPENDIX 1

Material examined. The number preceding the repository is the number of specimens; the following abbreviations are used for names of some of the collectors: DLG (D. L. Gustafson), GSS (G. Stace-Smith), HBL (Hugh B. Leech), MHH (Melvin H. Hatch), nc (no collector), RDK (Rex D. Kenner), and RSZ (Richard S. Zack).

CANADA.

BC: Abbotsford, roadside ditch, 14-ix-45, HBL, 3 CAS, 1 CNC; Chilliwack, 23, 27, 28-v-63, D.J. Farish, 5 UBCZ; Coquitlam, ditch behind warehouses at W end of Rocket Way, 27-ix-2004, RDK, 2 RDKC; Creston, Goat Mt. Lake, 5000 ft, 2-vii-33, GSS, 1 UBCZ; Creston, Goat River, 25-viii-46, GSS, 1 UBCZ; Creston, King Creek, 27-vii-48, GSS, 1 UBCZ; same loc., 8-ix-48, GSS, 2 UBCZ; same loc., 7, 15-vii-49, GSS, 3 UBCZ; same loc., 18-ix-55, GSS, 2 UBCZ; same loc., 19-ix-55, GSS, 1 INHS, 1 UBCZ; same loc., 21-ix-55, GSS, 1 UBCZ; same loc., 2-x-55, GSS, 1 UBCZ; Creston, 24-ix-55, GSS, 1 CAS, 1 CNC; Fernie, 31-viii-35, HBL, 3 CAS; Kitchener, roadside pond, 1-x-55, GSS, 1 UBCZ; Mission City, 18, 20-vi-53, G.J. Spencer, 2 CNC; Salmon Arm, 2-ix-29, HBL, 1 CAS; Surrey, shallow pond at 116 Avenue x 136 Street, 2-vi-2004, RDK, 3 RDKC; Surrey, extension of 110 Avenue N of 168 Street, 2-vi-2004, RDK, 2 RDKC; same loc., 3-ix-2004, RDK, 24 RDKC; Wynndel, head of Lizard Creek, 7-x-45, GSS, 3 UBCZ; same loc., 7-iv-46, GSS, 1 OSAC, 8 UBCZ; same loc., 23-vi-46, GSS, 1 UBCZ; same loc., 4-v-47, 1 UBCZ; same loc., 11-v-47, GSS, 14 CAS, 1 CNC, 8 UBCZ; same loc., 28-viii-47, GSS, 1 CNC, 2 UBCZ.

USA.

CA: LASSEN Co.: Norval Flats, 5500 ft, 15-ix-20, J.O. Martin, 90 CAS.

ID: BEAR LAKE Co.: Little Spring, below Davis Canyon campground, 18-viii-2004, S.M. Clark & R.W. Baumann, 5 BYU. BENEWAH Co.: Charlies Creek, 4-5 mi SE of Emida, 2-ix-86, RSZ, 1 WSU; E Fork of Charlies Creek, ca. 6 mi SE of Emida, 29-vii-87, RSZ, 51 WSU. BONNER Co.: Pack River, 8 mi N of Sandpoint, 22-ix-69, J. Schuh, 2 AMNH; Sagle, 4-vii-49, N.M. Downie, 1 OSAC. BUTTE Co.: Little Lost River, 10.6 mi N of Howe, 25-ix-91, RSZ, 1 WSU. CLEARWATER Co.: Badger Meadows ca. 7 mi E of Bovill, 28-viii-87, RSZ & V.L. Zack, 45 WSU. LATAH Co.: Big Meadow Creek Recreation Area, 5 mi. NW of Troy, 9-iv-87, RSZ, 1 WSU; E Fork Emerald Creek, ca 20 mi E of Harvard on Rt 447, 6-ix-90, RSZ, 1 WSU; N Fork of Palause River ca 11 mi NE of Harvard, 19-v-87, 2 WSU; pond by Rte 3, 7 mi SSW of Clarkia, 28-ix-90. LEMHI Co.: Canyon Creek, Railroad Canyon, 2.5 mi on Rte 29 below top of Bannock Pass, 7075 ft, 24-viii-69, HBL, 5 CAS. VAL-

LEY Co.: Trail Creek, FS Rte 22, 22.4 mi NNE of Cascade, 22-ix-91, RSZ, 3 WSU.

MT: DEER LODGE Co.: Jct Hwy 10A & Hwy 10, 7-viii-63, R.D. Anderson, 1 BYU. FLATHEAD Co.: Thompson R., Rte 56 ca 6.6 mi S of Rte 2, 25-ix-90, 1 WSU. GALLATIN Co.: no locality, 21-iv-24, nc, 3 MTEC; pond 2 mi from Bozeman, 17-v-55, nc, 2 MTEC; side ponds of Bridger Creek, 4800 ft, 28-vii-87, DLG, 3 MTEC; pond by Bridger Creek, 19-ix-87, DLG, 5 MTEC; Bridger Creek, 4800 ft, 26-iii-87, DLG, 3 MTEC; same loc., 18-v-87, DLG, 1 MTEC; same loc., 20-vi-87, DLG, 2 MTEC; Bridger Cr. 2 mi NE of Bozeman, 4800 ft, 18-v-86, DLG, 1 MTEC; Bridger Warm Springs, 3 mi NE of Bozeman on Hwy 86, 23-vii-89, C.B. Barr, 1 CBBC; E Gallatin River, 4600 ft, 22-vi-87, DLG, 1 MTEC; Gallatin River, 8 mi W of Bozeman, 23-viii-86, DLG, 3 MTEC; same loc., 1-ix-86, DLG, 3 MTEC; same loc., 13-x-86, DLG, 1 MTEC; Gallatin River, Bozeman, v–ix-88, DLG, pitfall trap, 2 MTEC; Gallatin River, weedy side pond, 4700 ft, 6-ix-87, DLG, 2 MTEC; same loc., 20-x-87, DLG, 1 MTEC; same loc. 10-xi-87, DLG, 4 MTEC; Gallatin R., 4700 ft, 12-iii-87, DLG, 7 MTEC; same loc., 25-v-87, DLG, 2 MTEC; same loc., 15-vii-87, DLG, 2 MTEC; same loc., viii-87, DLG, pitfall trap, 1 MTEC; same loc., 10-xi-87, DLG, 3 MTEC; same loc., 30-ix-88–12-iv-1989, DLG, pitfall trap, 3 MTEC; same loc., 12-iv–18-vii-89, DLG, pitfall trap, 2 MTEC. LAKE Co.: wet area just E of Swan River, ca 3 mi S of Swan L. at N.F. Road 129, 4-viii-89, C.B. Barr, 2 CBBC. LEWIS & CLARK Co.: Beaver Creek, 3-x-86, DLG, 1 MTEC; Beaver Creek near mouth, 3-x-86, DLG, 1 MTEC. LINCOLN Co.: Libby Fish Hatchery, 19-ix-86, DLG, 4 MTEC; same loc., 20-iii-87, DLG, 2 MTEC; same loc., 7-iv-88, DLG, 4 MTEC. MEAGHER Co.: *Sphagnum* bog & beaver pond with ice, 26.8 mi N of White Sulphur, 27-x-73, R.E. Roughley & M.L. Roughley, 1 JBWM. RAVALLI Co.: Lee Metcalf N.W.R., Pond 2, 9-viii-94, DLG, USFWS bottle trap, 1 MTEC; same loc., Pond 3, 9-viii-94, DLG, USFWS bottle trap, 3 MTEC; same loc., 8-ix-94, DLG, USFWS bottle trap, 2 MTEC; same loc., 30-ix-94, DLG, USFWS bottle trap, 2 MTEC.

OR: BAKER Co.: Beecher Creek, 7 mi N of Halfway, 4250 ft, 14-x-69, D. Gray, K. Gray, R. Rosenstiel, J. Schuh & D. Johnson, 1 AMNH; Richland "Env.", 2000 ft, 1-viii-45, H.P. Chandler, 4 EMEC. GRANT Co.: pond 9 mi W of Seneca, 15-x-71, J. Schuh, 1 AMNH; Rte 395 ca 9 mi N of Mt. Vernon, beaver pond, 17-vii-90, RSZ, 4 WSU; spring at SE Corner, 14-x-67, J. Schuh, 1 AMNH. MULTNOMAH Co.: marsh near Ainsworth State Park, 10-x-88, RSZ & R.D. Akre, 13 WSU. UNION Co.: Elgin, 29-viii-32, MHH, 1 OSAC.

UT: CACHE Co., A.J. Park, Blacksmith Fork Canyon, 14-vi-2003, M.J. Peterson, 3 BYU; Logan, 27-iv-63, E. Drake, 1 BYU; Spring Hollow, Logan Canyon, 16-iv-82, E. Coombs, 3 BYU; Spring Hollow pond, Logan Canyon, 5-v-89, nc, 6 BYU.

WA: CHELAN Co.: Rte 207 ca 3 mi N of Coles Corner, 13-ix-90, RSZ, 1 WSU. CLALLAM Co.: Matriotti Creek, Rte 101, 2.5 mi W of Sequim, 24-vi-92, RSZ, 2 WSU. CLARK Co.: E Fork of Lewis River ca 6 mi E of Heisson, 13-viii-89, J. Back, 1 WSU; Heisson ca 5 mi NE of Battle Ground, pools, 18-xi-88, J. Back, 5 WSU. GRAYS HARBOR Co.: Copalis Lagoon, 20-vii-33, T. Kincaid, 1 OSAC. JEFFERSON Co.: Quilcene, 26-vii-36, nc, 6 OSAC. KING Co.: Canyon Park, Bothell, 17-v-28, T. Kincaid, 2 OASC; pool ca 1.5 mi E of Redmond, 24-viii-89, RSZ, 1 WSU; Cedar Mt., 18-v-37, nc, 1 OSAC; same loc., 12-v-39, MHH, 4 OSAC; same loc., 6-vii-39, I.M., 2 OSAC; same loc., 9-v-40, MHH, 3 OSAC; same loc. & date, R.H. Foster, 2 OSAC; same loc., 10-vi-40, R.H. Foster, 2 OSAC; same loc., 22-v-41, MHH, 2 OSAC; same loc. & date, Thomas, 3 OSAC; same loc. & date, D.R. Orcutt, 3 OSAC; same loc., 26-iii-44, nc, 2 OSAC; same loc., 29-v-45, MHH, 2 OSAC; same loc., 16-v-46, MHH, 3 OSAC; same loc., 15-v-47, MHH, 3 OSAC, nc, 2 OSAC; Evans Creek, 30-viii-29, MHH, 1 OSAC; Malony's Grove, 20-iv-32, nc, 2 OSAC; North Bend, Malony's Grove, 20-ix-29, MHH, 14 OSAC; same loc., 10-v-30, MHH, 3 OSAC; same loc., 16-v-30, P. Ludy, 1 OSAC; same loc., 16-v-31, MHH, 3 OSAC; Redmond, 4-vi-67, D. Frechin, 1 WSU; Renton, 22-v-41, Campell, 1 OSAC; Renton, Cedar River, 22-v-41, Campell, 1 OSAC; same loc., 29-x-45, H.J. Jensen, 5 EMEC; pond in Forsgren Park, Seattle, 23-iv-92, K.A. Rosema, 4 WSU; Seattle, viii-28, nc, 1 OSAC; Snoqualmie Falls, 10-v-30, nc, 1 OSAC; Snoqualmie R., Malony's Grove, 13-v-28, MHH, 48

OSAC; Stillwater, 11-ii-34, nc, 1 OSAC. KITTITAS Co.: Rte US 10, 2.9 mi NW of Ellensburg, 8-x-77, R. Thorne, 1 WSU; Easton, 28-iv-39, MHH, 1 OSAC; Ellensburg, 19-vii-32, MHH, 2 OSAC; Jungle Cr., FS Rte 19 ca 11 mi N of Cliffdell, 31-viii-90, RSZ, 1 WSU; Manastash Canyon, ca 10 mi WSW of Ellensburg, beaver pond, 3-xi-88, RSZ, 1 WSU; Roslyn Ponds, 7-v-2000, E. Sugden, 1 BYU. KLICKITAT Co.: pool by Klickitat River ca 5.5 mi N of Lyle, 17-viii-89, RSZ, 1 WSU; Outlet Creek ca 1.5 mi S of Glenwood, 17-viii-89, RSZ, 2 WSU. MASON Co.: irrigation ditch 3.5 mi SW of Kamilche on Rte 108, 19-i-94, RSZ, 1 WSU. OKANOGAN Co.: Granite Creek ca 6 mi W of Republic on Rte 20, 11-ix-90, RSZ, 1 WSU; small pool 12 mi N of Nespelem on RTE 155, 30-ix-87, R.D. Akre, 23 WSU. PIERCE Co.: Mt. Rainier National Park, Longmire, Fish Creek Valley, spring-fed pond, 3000 ft, 15-vi-69, I. Smith, 36 ROME; Mt. Rainier National Park, Longmire, Fish Creek Valley, shallow pond in flood plain, 3000 ft, 15-vi-69, I. Smith, 10 ROME; Mt. Rainier National Park, Tahoma Creek, 2300 ft, 12-viii-73, A. Smetana, Z. Smetana & D. Smetana, 2 CNC; WSU Res & Ext. Center, Puyallup, pond, 24-ix-92, RSZ, 2 WSU. SKAMANIA Co.: Moffett Creek ca 2 mi W of North Bonneville, 12-x-88, RSZ & R.D. Akre, 16 WSU. SNOHOMISH Co.: Canyon Park, 15-x-28, nc, 2 OSAC; same loc., 6-v-30, nc, 2 OSAC; same loc., 8-vii-32, nc, 1 OSAC; Martha Lake, 5-v-31, nc, 1 OSAC; Monte Cristo, in stomach of rainbow trout, 987 m, 9-vii-38, Kiser, 1 OSAC; Sultan, 1-vi-53, B. Malkin, 1 OSAC; Thomas Lake, 5-vii-32, nc, 1 OSAC; same loc., 31-v-34, nc, 1 OSAC; same loc., 10-vii-34, nc, 3 OSAC. WHATCOM Co.: Kendall Creek, 14-viii-32, nc, 1 OSAC; Lynden, 6-vii-64, nc, 3 OSAC; same loc., 7-viii-64, nc, 3 OSAC; same loc., 2-vii-65, L. Russell, 4 OSAC; N Fork Nooksack River, 17-vii-32, nc, 1 OSAC; Silver Lake, 14-viii-32, nc, 1 OSAC. YAKIMA Co.: Milk Creek, FS Rte 12, 1-viii-90, RSZ, 2 WSU.

WY: TETON Co.: Snake River, 15.8 mi S of Jackson on Rte 26/89, 24-ix-91, RSZ, 1 WSU.

Lestes disjunctus and *L. forcipatus* (Odonata: Lestidae): An evaluation of status and distribution in British Columbia

ROBERT A. CANNINGS[1] and JOHN P. SIMAIKA[2]

ABSTRACT

Of the five species of the damselfly genus *Lestes* that live in British Columbia, *Lestes forcipatus* Rambur and *L. disjunctus* Selys are the most difficult to separate morphologically. Females can be readily distinguished by the size of the ovipositor, but males are difficult to separate. In British Columbia, *L. disjunctus* is more common, widespread and familiar. Before 1998, when it was first reported in BC, specimens of *L. forcipatus* were misidentified as *L. disjunctus* because the former is known mainly from eastern North America and most *Lestes* species are usually most readily identified using male characters. The identities of museum specimens of the two species were checked and corrected by us as necessary. Ecological and behavioural observations and up-dated distribution maps of the species are presented. Throughout its range in BC, *L. forcipatus* is mostly sympatric with *L. disjunctus* but lives in a narrower range of habitats and localities – mostly cool sedge marshes and fens. The two species show some temporal and behavioural separation.

Key Words: Odonata, *Lestes disjunctus*, *Lestes forcipatus*, British Columbia, distribution, habitat preference, plant associations, temporal separation, oviposition

INTRODUCTION

Five species of *Lestes* occur in British Columbia (BC): *L. congener* Hagen (spotted spreadwing), *L. disjunctus* Selys (northern spreadwing), *L. dryas* Kirby (emerald spreadwing), *L. forcipatus* Rambur (sweetflag spreadwing), and *L. unguiculatus* Hagen (lyre-tipped spreadwing). *Lestes disjunctus* is the most common, widespread and familiar of these in the province, and one of the most abundant odonates in Canada where it ranges as far north as the Arctic treeline (Cannings 2002). It inhabits many types of standing water habitats with abundant aquatic vegetation and, in southern BC, adults have been recorded from mid-June to mid-October (Cannings 2002).

Lestes forcipatus, although as abundant as *L. disjunctus* in some cold fen habitats, is generally much less common; both species often occur at the same site. *Lestes forcipatus* does not range as far north as *L. disjunctus* but, although not known from much of BC's north, it has been collected in south-eastern Yukon (S.G. Cannings, pers. comm.). In the western Canadian Cordillera, it is most common in sedge fens (Cannings 2002). Walker (1953) described *L. forcipatus* habitat in Ontario as "ponds, both temporary and permanent, marshy lakes, and slow, weedy streams". In BC, adults of *L. forcipatus* have been reported from mid-May to mid-September (Cannings 2002).

Lestes forcipatus was first recorded in BC in 1998, when it was collected in the Rocky Mountain Trench north of Golden and subsequently found in many other localities in the south-eastern part of the province (Cannings *et al.* 2005). However, undoubtedly it has long been a resident of the province but was overlooked because of its close resemblance to *L. disjunctus*. Before 1998, *L. forcipatus* was unknown west of Saskatchewan in Canada (Walker 1953, Westfall and May 1996), although in 1997

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it was found in Washington State, the first record west of Montana in the United States (Cannings *et al.* 2005). The species is now known from seven counties in Washington and one in Idaho (Paulson 2005). Subsequently, collectors found *L. forcipatus* at several other BC locations farther south and west in 1999; by 2000 it had been collected on Vancouver Island. Inventories in northern BC (2000-2003) have extended its range to about 55ºN latitude (Cannings *et al.* 2005), although records in south-eastern Yukon in 2004 and 2005 (S.G. Cannings, pers. comm.) indicate it probably ranges through much of north-eastern BC, at least. In addition, Catling *et al.* (2004) reported the species from southern Northwest Territories near Fort Smith. A map of the distribution of *L. forcipatus* in North America was published by Donnelly (2004).

Catling (2002), Donnelly (2003) and Simaika and Cannings (2004) provided practical information on characters for the identification of the two species. Simaika and Cannings (2004) particularly emphasized the usefulness of pruinosity patterns in western North American populations. A recent check of some specimens in the Royal BC Museum (RBCM) (Victoria) and the Spencer Entomological Museum, University of BC (SEM) (Vancouver) by one of us (JPS) revealed that a signficant number identified as *L. disjunctus* (even among those collected after 1998) are misidentified *L. forcipatus* (Cannings *et al.* 2005). The discovery of misidentified specimens indicates that other museum collections across western Canada probably contain such specimens. Herein we report the results of an identification check of all *L. disjunctus* specimens in the RBCM and the SEM, the two collections holding the majority of Odonata specimens from BC.

This paper establishes accurate distributions for both species in BC by publishing up-dated distribution maps. We also provide revised information on habitat preferences and life histories.

MATERIALS AND METHODS

Specimens. We examined 1853 specimens previously identified as *L. disjunctus* in the RBCM and SEM collections and separated the two species using characters documented in Simaika and Cannings (2004). Females were identified by the relative lengths of the ovipositor. The most useful character for separating males is the amount of pruinescence on the thorax and, in *L. forcipatus*, the presence of a bare, non-pruinose patch on the posterior third of the dorsum of the second abdominal segment.

Habitat and life-history data. Information on habitat preferences, emergence times, flight period and breeding behaviour was extracted from the RBCM and SEM databases. The wetland site association classification used is that of MacKenzie and Moran (2004); site associations noted are listed and defined in Table 1.

Distribution maps. Maps (Figs. 1 and 2) were produced electronically from the databases of the RBCM and SEM by Clover Point Cartographics Ltd. (Victoria, BC) using Microsoft Visual Basic version 6 and Environmental Systems Research Institute (ESRI) Arc Info Workstation version 9.0 (ESRI 2005). The base line features are from Terrain Resource Information Mapping (TRIM) 1: 2,000,000 and the surface model is based on Clover Point and TRIM Digital Elevation Model data. TRIM data are used under license from the BC Ministry of Environment.

RESULTS AND DISCUSSION

Changes in specimen identification. In the RBCM collection, 38 specimens (23♂, 15♀) from British Columbia collected before 2004 and previously identified as *L. disjunctus* were re-identified as *L. forcipatus*; five (3♂, 2♀) identifications were changed in the SEM collection. These changes represent 2.3% of the sample. This material came from ten localities over the southern two-thirds of the province in the

Table 1.

Main wetland habitat types used by *Lestes disjunctus* (*L. dis.)* and *L. forcipatus* (*L. for.*) in British Columbia. Wetland site associations and codes are taken from MacKenzie and Moran (2004). Site association names indicate dominant plant species used to define the habitat type; codes are used in the discussion of *Lestes* habitat in the text. A, absent; C, common; R, rare; U, uncommon.

Ecosystem Type	Association Code	Site Association Name	*L. dis.* status	*L. for.* status
Saline associations at grassland ponds	Gs01	*Distichlis spicata* var. *stricta* (Alkali saltgrass)	C	A
	Gs02	*Puccinellia nuttalliana – Hordeum jubatum* (Nuttall's alkali-grass - Foxtail barley)	C	A
	Gs03	*Carex praegracilis* (Field sedge)	C	A
Bogs	Wb12	*Scheuchzeria palustris – Sphagnum* (Scheuchzeria – Peat-moss)	U	U
	Wb13	*Carex limosa – Menyanthes trifoliata – Sphagnum* spp. (Shore sedge - Buckbean - Peat-moss)	U	A
	Wb50	*Ledum groenlandicum – Kalmia microphylla – Sphagnum* spp. (Labrador Tea – Bog-laurel - Peat-moss)	C	A
	Wb51	*Pinus contorta – Empetrum nigrum – Sphagnum austinii* (Shore pine–Black crowberry–Tough peat-moss)	C	A
	Wb52	*Juniperus communis – Trichophorum cespitosum – Rhacomitrium lanuginosum* (Common juniper – Tufted clubrush – Hoary rock-moss)	C	A
Fens	Wf01	*Carex aquatilis – Carex utriculata* (Water sedge – Beaked Sedge)	C	U
	Wf02	*Betula nana – Carex aquatilis* (Scrub birch – Water sedge)	C	U
	Wf03	*Carex aquatilis – Sphagnum* (Water Sedge – Peat-moss)	R	R
	Wf04	*Salix barclayi – Carex aquatilis – Aulacomnium palustre* (Barclay's willow – Water sedge – Glow moss)	U	R
	Wf05	*Carex lasiocarpa – Drepanocladus aduncus* (Slender sedge – Common hook-moss)	C	U
	Wf06	*Carex lasiocarpa – Menyanthes trifoliata* (Slender sedge – Buckbean)	C	C
	Wf07	*Betula nana – Menyanthes trifoliata – Carex limosa* fens (Scrub birch – Buckbean – Shore sedge)	C	C
	Wf08	*Carex limosa – Menyanthes trifoliata – Drepanocladus* spp. (Shore sedge – Buckbean – Hook moss)	C	C
	Wf09	*Eleocharis quinqueflora – Drepanocladus* (Few-flowered spike-rush – Hook moss)	U	A
	Wf10	*Trichophorum alpinum – Scorpidium revolvens* (Hudson Bay clubrush – Red hook-moss)	C	C
	Wf12	*Eriophorum angustifolium – Caltha leptosepala* (Narrow-leaved cotton-grass – Marsh-marigold)	C	A
Marshes	Wm01	*Carex utriculata – Carex aquatilis* (Beaked sedge – Water sedge)	C	U
	Wm02	*Equisetum fluviatile - Carex utriculata* (Swamp horsetail – Beaked sedge)	C	U
	Wm04	*Eleocharis palustris* (Common spike-rush)	C	R
	Wm05	*Typha latifolia* (Cattail)	C	R
	Wm06	*Schoenoplectus acutus* (Great bulrush)	C	R
	Wm07	*Juncus balticus* (Baltic rush)	C	A

Figure 1. Distribution of records of *Lestes disjunctus* in British Columbia to 2004. Data represent specimen records only from the Royal BC Museum, Victoria, and the Spencer Entomo-

vicinities of Bowser, Cawston, Duncan, Fort St. James, Germansen Landing, Horsefly, Mackenzie, Qualicum Beach, Rogers Pass, and Wells Gray Provincial Park.

Distribution and status. The newly plotted range maps for *L. disjunctus* (Fig. 1) and *L. forcipatus* (Fig. 2) update our knowledge of the BC distributions of the species to the 2004 collecting season. The distribution of *L. disjunctus* in the province remains unchanged; it is so common and widespread that the subtraction of specimens from ten localities made little difference to the updated range map (Fig. 1). Indeed, in all but a very few localities, *L. disjunctus* has been collected wherever *L. forcipatus* is found. *Lestes disjunctus* is second only to *Enallagma boreale* Selys as the most frequently collected odonate in BC; it occurs over the entire province where dragonflies are able to live; the only major gaps in its known distribution are those areas difficult to access by road.

Although our reidentifications show that *L. forcipatus* has lived in the region since long before 1998, all but five of the 43 known localities were recorded from 1998 to 2004. Thus, in terms of its known status in BC, the species has gone from anonymity to being a widespread and fairly common taxon in only seven years. Although it was collected at two localities in the year of its discovery and 12 the year after, it was retained on the provincial Blue List of species of concern until 2004 (Ramsay and Cannings 2005). Despite this rapid change in its status, *L. forcipatus* is clearly less common and widespread than *L. disjunctus*

Figure 2. Distribution of records of *Lestes forcipatus* in British Columbia to 2004. Data represent specimen records only from the Royal BC Museum, Victoria, and the Spencer Entomological Museum, University of BC, Vancouver.

in BC. In the Okanagan-Similkameen basin, probably the most thoroughly collected area of the province, it has been found at only one locality (near Cawston). It has yet to be collected in the Shuswap region or in the Lower Mainland of south-eastern BC and still must be considered rare on the coast in general. It was not found in intensive surveys in the Peace River and Fort Nelson regions in 1997, nor along Highway 37 or in the Atlin area in 2003, although in 2004 and 2005 it was collected in south-eastern Yukon (S.G. Cannings, pers. comm.). Further study will likely fill some of these gaps; nevertheless, the narrower range of its preferred habitats will continue to make it harder to find than *L. disjunctus* in most places in BC.

Habitat Requirements. See Table 1. A major basis for the greater abundance and wider distribution of *L. disjunctus* compared with *L. forcipatus* is the ability of the former to use a wider range of habitats. This is especially true of warmer habitats in the southern parts of the province such as eutrophic marshes (Wm04-07) and saline ponds (Gs01-03), where *L. disjunctus* is common and *L. forcipatus* is rare or absent. *Lestes forcipatus* has yet to be found in the widespread bogs of the outer coast, from the Queen Charlotte Islands and Prince Rupert regions to Vancouver Island and the Fraser River delta (Wb13, Wb50-52). Both species are found in *Carex* and *Equisetum* marshes (e.g. Wm01-02), but *L. disjunctus* is more common in these places. *Lestes forcipatus* appears to be most frequent in fens or bogs dominated by *Carex*,

Trichophorum, *Menyanthes, Comarum* and mosses such as *Sphagnum*, *Drepanocladus* and *Scorpidium* (Wb12, Wf01-10, 12). However, *L. disjunctus* is usually more abundant in these habitats and, apparently, *L. forcipatus* is absent from many localities with such habitat types, especially in the North, that superficially appear ideal for its development. Walker (1953) notes that, in eastern Canada, *L. forcipatus* is also more locally distributed than *L. disjunctus*. In summary, *L. forcipatus* is most common in cold sedge and moss fens and uncommon, rare or absent in warmer habitats such as eutrophic marshes.

Life histories. *Lestes disjunctus* adult records range from 15 May to 9 October. The bulk of them fall between late June and early August with a peak in the last half of July. For example, on 22 July 1996 hundreds of teneral adults were observed at Burns Bog in the Fraser River delta. Records of mating pairs range from 12 July to 5 October and oviposition dates range from 12 July to 17 September. Adult records of *L. forcipatus* range from 18 May to 4 September; about 85% of these are from late June through late July, with the peak in the last half of July. Mating has been observed from 28 June to 14 August and oviposition from 12 July to 14 August. Although there is strong overlap of the flight periods of the two species in BC, there is some evidence that adult *L. forcipatus* emerge earlier than *L. disjunctus* where they co-occur. At Nahlbeelah wetlands near Kitimat on 10 July 2005, fully mature adult *L. forcipatus* were common but the population of *L. disjunctus* was just beginning to emerge. At Hamilton Marsh near Qualicum Beach, adult *L. forcipatus* were flying on 18 May 2004; adult *L. disjunctus* appeared on 23 June and sexually mature specimens were not observed until 3 July. This suggests a difference of about two to four weeks in emergence times of the two species and is similar to the amount of time between the first emergence of the two species in eastern Canada noted by Walker (1953).

In addition to a possible temporal shift in the flight period and, consequently, the mating times of the two species, there may be some interspecific differences in oviposition behaviour. Simaika (2005) and Simaika and Cannings (2006) reported that at Hamilton Marsh, near Qualicum Beach, ovipositing females of *L. disjunctus* inserted eggs into only two species, *Carex lanuginosa* Michaux and *Juncus arcticus* Willdenow. Females on *C. lanuginosa* oviposited into fresh stems, just above the water surface; on *J. arcticus* they laid eggs in dead stem tissue, about 10 cm from the tip of the stem. *Lestes forcipatus* will also oviposit on *C. lanuginosa* and *J. articus* but, unlike *L. disjunctus*, it appears to prefer the living stems of *J. arcticus* and will also utilize *Menyanthes trifoliata* L.

These observations suggest that there may be some niche separation of *L. disjunctus* and *L. forcipatus* in BC. More research is required to elucidate the ecological and behavioural differences between these two closely related, sympatric damselflies.

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Mammal Fleas (Siphonaptera: Ceratophyllidae) New for Alaska and the Southeastern Mainland Collected During Seven Years of a Field Survey of Small Mammals

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ABSTRACT

Ten taxa of mammal fleas were among 124 collection records from 12 host species (one shrew, nine rodents and two carnivores), at 72 localities on the southeastern Alaska mainland in 1989 and during an extensive survey of mammals in 1992-1995 and 1997-1999. *Megabothris asio megacolpus* (Jordan) ex *Microtus pennsylvanicus* (Ord), *Malaraeus telchinus* (Rothschild) ex *Peromyscus keeni* (Rhoads) and *Clethrionomys gapperi* (Vigors) are new fleas for Alaska. *Orchopeas caedens* (Jordan) ex *Tamiasciurus hudsonicus* (Erxleben) is a new flea for southeastern Alaska. *Synaptomys borealis* (Richardson) is a new host record for *Opisodasys k. keeni* (Baker). The other six taxa of fleas collected were *Hystrichopsylla dippiei spinata* Holland, *H. o. occidentalis* Holland, *Catallagia charlottensis* (Baker), *Ceratophyllus ciliatus protinus* Jordan, *Megabothris abantis* (Rothschild) and *Opisodasys vesperalis* (Jordan). Of these, *H. o. occidentalis*, *C. charlottensis* and *M. abantis* have seven new host records for the southeastern Alaska mainland. Distribution patterns of the fleas and their host relationships in North America are discussed.

Key Words: fleas, Siphonaptera, mammals, Alaska

INTRODUCTION

The advancement in knowledge of the fleas of southeastern Alaskan mammals has lagged behind that of Alaska west of the Yukon Territory in part due to difficulties of travel in the fragmented, rugged coastal to montane topography. An extensive survey by the University of Alaska Museum, Fairbanks, of shrews, mice, voles, lemmings and some larger mammals, such as arboreal squirrels during 1992-1995 (MacDonald and Cook 1996) and 1997-1999 included the collection of fleas. This survey produced 124 collection records (including three from an earlier study of marten) at 72, mostly new, localities. Two fleas of mice and voles new for Alaska, one squirrel flea new for southeastern Alaska, seven new host records for three other fleas for the southeastern Alaska mainland, and one new lemming host record for a mouse flea were added (Table 1).

MATERIALS AND METHODS

The fieldwork for the mammal survey was conducted as described by Murrell *et al.* (2003) on ticks collected from some of the same mammal specimens that produced some of the fleas reported on here. Full data for the mammal specimens can be obtained at http://arctos.database.museum by tracking the University of Alaska Museum of the

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Table 1.
Mammalian hosts of the 10 taxa of fleas with present records for the southeastern Alaska mainland.

Mammal	Fleas
Sorex cinereus Kerr, masked shrew	*Hystrichopsylla o. occidentalis* Holland[1]
Tamiasciurus hudsonicus (Erxleben), red squirrel	*Ceratophyllus ciliatus protinus* Jordan *Orchopeas caedens* (Jordan)[2]
Glaucomys sabrinus (Shaw), n. flying squirrel	*Opisodasys vesperalis* (Jordan)
Peromyscus keeni (Rhoads), Keen's mouse	*H. o. occidentalis* *Catallagia charlottensis* (Baker) *C. c. protinus* *Megabothris abantis* (Rothschild) *Opisodasys k. keeni* (Baker) *Malaraeus telchinus* (Rothschild)[3]
Clethrionomys rutilis (Pallas), n. red-backed vole	*H. o. occidentalis*[1] *C. c. protinus* *M. abantis*
C. gapperi (Vigors), s. red-backed vole	*H. o. occidentalis*[1] *C. charlottensis* *M. abantis*[1] *M. telchinus*[3]
Microtus pennsylvanicus (Ord), meadow vole	*C. charlottensis*[1] *M. abantis*[1] *Megabothris asio megacolpus* (Jordan)[3]
M. longicaudus (Merriam), long-tailed vole	*M. abantis*
Synaptomys borealis (Richardson), n. bog lemming	*M. abantis*[1] *O. k. keeni*[4]
Zapus hudsonius (Zimmerman), meadow jumping mouse	*M. abantis*
Martes americana (Turton), marten	*Hystrichopsylla dippiei spinata* Holland
Mustela vison Schreber, mink	*H. d. spinata*[5]

[1] New host record for southeastern Alaska.
[2] New for southeastern Alaska.
[3] New for Alaska.
[4] New host record.
[5] Probably from marten.

North AF number listed under Material Examined.

In the laboratory the fleas were prepared for microscopic study by transferring them from the labeled field vials of 70% ethanol to a rinse in distilled water, then submerged in 10% KOH until sufficiently bleached (1 hr to 3 days), rinsed two or three times in distilled water, dehydrated in graduated ethanols (to 90%), degreased in oil of wintergreen, rinsed in xylene and mounted in Canada balsam on labeled microscope slides. Voucher specimens were deposited in the United States National Museum (USNM) and the Canadian National Collection of Insects and Arachnids (CNC). Following is a list of collectors and their acronyms used in this paper: C. J. Conroy (CJC), J. A. Cook (JAC), J. Foreit (JF), R. Heinen (RH), S. O. MacDonald (SOM), S. R. Peterson (SRP), A. M. Runck (AMR), C. T. Seaton (CTS), K. D. Stone (KDS), A. A. Tsvetkova (AAT) and M. J. Wike (MJW). Specimens without acronyms are in the collections of the authors.

SPECIES ACCOUNTS

HYSTRICHOPSYLLIDAE

Hystrichopsylla dippiei spinata Holland, 1949

Material examined: USA: AK: all from Juneau area; 30.4 km NW, between Amalga Harbor and Windfall Lake, 1♀ ex *Martes americana* (Turton), 13-xii-89, SRP; ca. 34 km NW, Yankee Basin trail, 1♀ same host, 30-xii-89, SRP; Eagle River trail, 1♀ ex *Mustela vison* Schreber or *Martes americana*, 30-xii-89, SRP.

These three records continue a series begun in 1987 with the first record reported by Haas *et al.* (1989). The range of this large flea was only extended ca. 3.3 km farther NW of Juneau. Although most hystrichopsyllids are associated with insectivores or rodents, this flea infests mustelids. The new records provide support for marten as the true host. The Eagle River mink and marten were packed together in the same container. Only one valid record (1♀) for mink (Haas *et al.* 1979) exists. Therefore, the original host for the Eagle River record may have been the marten. Thus, 24 specimens (8♂♂, 16♀♀) and 18 collection records (Haas *et al.* 1978, 1979, 1980, 1982, 1989, present study) in southeastern Alaska including islands are now known. Except for one mink, two humans, and the uncertain host record noted above, all host specimens were marten. There are no Alaskan records from shrews or rodents although such collections have been made in British Columbia and Oregon (Holland 1957; Hopkins and Rothschild 1962; Lewis *et al.* 1988). Most specimens were from skunks (*Spilogale* spp.). Holland (1949) also had two females from marten and ermine (*Mustela erminea*) from Vancouver Island that he excluded from the series of *H. spinata* new species.

Hystrichopsylla occidentalis occidentalis Holland, 1949

Material examined: USA: AK: Berg Bay, 56°21'49"N, 132°00'29"W, 1♀ ex *Clethrionomys gapperi* (Vigors) [AF 21819], 4-viii-97, CTS. Echo Cove, 59° 31'45"N, 134°21'58"W, 1♂ ex *Clethrionomys rutilis* (Pallas) [AF21587], 16-vii-97, CTS. Frosty Bay, S side, 56°03'28"N, 131° 58'01"W, 1♀ ex *C. gapperi* [AF22904], 8-viii-97, CTS. Klukwan, 17 km W & 30 km N, Kelsall River drainage, 1♂, 1♀ ex *Sorex cinereus* Kerr [AF8096], 30-vii-94, JAC, SOM. Taku River, Canyon Island, 58° 33'N, 133°41'W, 1♀ ex *Peromyscus keeni* (Rhoads) [AF8272], 17-vii-94, JAC; same data but *C. gapperi* [AF2856], 16-vii-94, JAC.

This small relative of *H. d. spinata* is a hygrophilous parasite of shrews, voles (type host: *Clethrionomys gapperi*) and mice and occurs in a long narrow range with a noticeable concentration of collection localities along the coast from northern California to southwestern Alaska (Holland 1949: Map 6; Campos and Stark 1979: Fig. 60; Holland 1985: Map 12; Lewis *et al.* 1988: p. 63). The six new mainland records for five new localities as well as the four earlier records indicate that this flea is uncommon on trapped hosts, in accordance with its behaviour like a nest flea. Consequently, the majority of the fleas reside in nests of the hosts. For example, from two nests of *Microtus oeconomus* (Pallas) on the Chilkat Peninsula, Haas (1982) collected a total of 5♂♂ and 4♀♀ *H. o. occidentalis*. This exceeds the total of 2♂♂ and 5♀♀ from the six trapped host specimens listed above. The predominance of nest populations was confirmed for *H. occidentalis linsdalei* Holland with large samples of nests (179) and hosts (877) during a 2.5-y survey in northern California by Stark (2002). He reported a seasonal association with higher populations in nests of voles than on trapped voles except during a short time in fall.

Sorex cinereus is a new host record for southeastern Alaska. Haas *et al.* (1980) listed *Sorex monticolus* Merriam (as *Sorex vagrans* Baird - see MacDonald and Cook 1996) as a host of *H. o. occidentalis* near Yakutat. The two species of red-backed voles, *C. gapperi* and *C. rutilis*, are also new host records for *H. o. occidentalis* in southeastern Alaska.

CTENOPHTHALMIDAE

Catallagia charlottensis (Baker, 1898)

Material examined: Hosts were *Peromyscus keeni* unless otherwise indicated. USA: AK: Gwent Cove, 54°57'00"N, 130° 20'00"W, 1♀ [AF26560], 17-viii-98, SOM. Haines, Chilkoot Lake, 59°18'39"N, 135° 34'02"W, 2♀♀ [AF4593], 11-vi-93, MJW. Klukwan, 5 km W of, Klehini River, 59° 24'39"N, 136°00'09"W, 1♀ ex *Microtus pennsylvanicus* (Ord) [AF8036], 29-vi-94, JAC. Mosquito Lake, 59°27'08"N, 136° 01'38"W, 2♀♀ [AF28822], 5-vi-99, AMR. Reflection Lake, W side, 56°00'33"N, 131° 34'32"W, 1♀ ex *Clethrionomys gapperi* [AF29075], 30-vi-99, AMR. Rudyerd Bay, Point Louise, 55°32'42"N, 130°52'13"W, 1♀ ex *C. gapperi* [AF29307], 10-vii-99, AMR. Rudyerd Bay, 55°33'16"N, 130° 51'33"W, 1♀ [AF29381], 12-vii-99, AMR. Smeaton Bay mouth, 55°18'09"N, 130° 50'38"W, 2♀♀ [AF29276], 9-vii-99, AMR. Taku River, Canyon Island, 58°33'N, 133° 41'W, 1♂ [AF8274], JAC; same data but 1♀ [AF8276]. Turner Creek, 58°10'40"N, 133°57'30"W, 1♀ [AF10126], 20-vii-94, SOM, JAC, CTS. Walker Cove, Ledge Point, 55°42'20"N, 130°53'34"W, 1♀ [AF29458], 14-vii-99, AMR.

The distribution patterns of this hygrophilous nest flea and of *H. o. occidentalis* are similar (Holland 1963: Fig 2, 1985: Map 14; Lewis *et al.* 1988: p. 82; Haas *et al.* 1989: Fig 2; Lewis and Haas 2001). In southeastern Alaska both fleas have been recorded from *P. keeni* (as *P. maniculatus*) and *M. oeconomus* (nests) at mainland localities (Haas 1982; Haas *et al.* 1982; Holland 1985). Our new records of these two fleas and a record of *C. charlottensis* ex *C. rutilis* in Haines (Holland 1985) linked the fleas as parasites of *C. rutilis*, *C. gapperi*, and *M. pennsylvanicus* on the mainland. Thus far however, only *C. charlottensis* is known from *M. longicaudus* in our survey area (Haas *et al.* 1982). Another similarity in our records for *H. o. occidentalis* and *C. charlottensis* from trapped hosts was the infrequency of more than a single specimen collected per host. The larger number of *H. o. occidentalis* specimens in nests of *M. oeconomus* than on all trapped hosts applies to *C. charlottensis* for its occurrence in the same two nests found along the shore of the Chilkat Peninsula. Both nests were infested with breeding populations from which were collected a total of 13♂♂ (four reared) and 11♀♀ (three reared); an additional male was collected from a third nest. Again, more specimens (25) were collected from nests (3) than those (15) from trapped hosts (12).

CERATOPHYLLIDAE

Ceratophyllus ciliatus protinus Jordan, 1929

Material examined: Hosts were *Tamiasciurus hudsonicus* (Erxleben) unless otherwise indicated. USA: AK: Berg Bay, 56° 21'49"N, 132°00'29"W, 4♂♂, 2♀♀ [AF21810], 2-viii-97, CTS. Chickamin River, Wolf Cabin, 55°46'N, 130°53'W, 1♂ [AF4930], 25-vii-93, SOM. Dyea National Historical Park, 59°30'24"N, 135° 20'52"W, 1♂, 2♀♀ [AF12532], 2-vii-95, CTS. Gwent Cove, 54°57'00"N, 130° 20'00"W, 2♂♂, 1♀ [AF26585], 19-viii-98, SOM. Klukwan, 5 km W, Klehini River, 59°24'39"N, 136°00'09"W, 1♂ [AF8117], 1-vii-94, JAC. Peterson Creek, Juneau Quad., 58°29'N, 134°47'W, 1♀ ex *Clethrionomys rutilis* [AF8243], 11-vii-94, JAC. Rudyerd Bay, 55°33'16"N, 130° 51'33"W, 2♀♀ [AF29409], 13-vii-99, AMR. Taku River, Canyon Island, 58° 33'N, 133°41'W, 1♂ ex *Peromyscus keeni* [AF8272], 17-vii-94, JAC; same data but 2♂♂ [AF8273]. Walker Cove, Hut Point, 55°42'48"N, 130°54'04"W, 1♂ ex *P. keeni* [AF29442], 13-vii-99, AMR.

This member of the Vancouverian group (Holland 1963) has a typical Northwest Pacific coast distribution similar to two other members of the group, *H. o. occidentalis* and *C. charlottensis* (Haddow *et al.* 1983: Map 17; Holland 1985: Map 71; Lewis *et al.* 1988: p. 179). These authors described the changes of preferred hosts along the coast from south to north with Townsend's chipmunk (*Neotamias townsendii* (Bachman)) in Oregon, Douglas's squirrel (*Tamiasciurus douglasii* (Bachman)) in southwestern British Colum-

bia, and the red squirrel (*T. hudsonicus*) in southeastern Alaska. Originally thought to be the only truly specific flea of the red squirrel in this area of Alaska, this view has been modified as the result of collection of the much wider-ranging true red squirrel flea, *Orchopeas caedens* Jordan, along the Taiya River in 1995 (see below).

Megabothris abantis (Rothschild, 1905)

Material examined: USA: AK: Bartlett Cove, 10 km NW of Gustavus Airport, 58° 27'N, 135°53'W, 1♂ ex *Clethrionomys rutilis* [AF2379], 16-vii-92, collector unknown. Berg Bay, 56°21'49"N, 132° 00'29"W, 1♀ ex *C. gapperi* [AF21819], 4-viii-97, CTS. Chickamin River, Wolf Cabin, 55°46'N, 130°53'W, 1♂ ex *Synaptomys borealis* (Richardson) [AF4973], 26-vii-93, SOM *et al.* Chilkat Peninsula, Mud Bay, 59°09'45"N, 135°21'28"W, 1♂, 1♀ ex *Clethrionomys rutilis* (2) [AF22019, 22020], 6-vii-97, CTS *et al.* Frosty Bay, S side, 56°03'28"N, 131°58'01"W, 1♀ ex *C. gapperi* [AF22904], 8-viii-97, CTS. Klukwan, 5 km W of, Klehini River, 59° 24'39"N, 136°00'09"W, 2♂♂, 1♀ ex *Zapus hudsonius* (Zimmerman) (2) [AF8068, 8070], 30-vi-94, JAC. Nakat Inlet, 54°57'N, 130°45'W, 1♀ ex *C. gapperi* [AF4265], 8-vii-93, JAC, SOM. Rudyerd Bay, 55°33'16"N, 130°51'33"W, 2♀♀ex *C. gapperi* [AF29315], 10-vii-99, AMR; same data but 1♀ [AF29375], 12-vii-99; same data but 1♀ [AF29408], 13-vii-99; same data but 55°41'58"N, 130° 31'12"W, 1♀ ex *P. keeni* [AF22589], 8-vi-99, RH, AMR. Rudyerd Bay, Point Louise, 55°32'42"N, 130°52'13"W, 4♀♀ ex *C. gapperi* (2) [AF29304, 29307], 10-vii-99, AMR. Salmon River, mouth of Texas Creek, 56°01'37"N, 130°04'14"W, 1♀ ex *P. keeni* [AF12736], 2-viii-95, CTS. Smeaton Bay mouth, 55°18'09"N, 130° 50'38"W, 1♂, 1♀ ex *C. gapperi* [AF29290], 10-vii-99, AMR; same date but 1♂ ex *P. keeni* [AF29292]. Stikine River, Figure 8 Lake, 56°42'N, 132°15'W, 1♀ ex *P. keeni* [AF2628], 14-vii-92, SOM; same data but [AF2650], 15-vii-92. Taku River, Canyon Island, 58°33'N, 133°41'W, 1♂ ex *C. gapperi* [AF8254], 16-vii-94, JAC; same data but 1♀ [AF8270], 17-vii-94; same data but 1♂, 2♀♀ [AF8271]; same data but 2♀♀ ex *M. pennsylvanicus* [AF8268], 16-vii-94. Turner Creek, 58°10'40"N, 133° 57'30"W, 2 ♀♀ ex *P. keeni* [AF10126], 20-vii-94, SOM, JAC, CTS; same data but 1♀ ex *C. gapperi* [AF10119]; same data but 1♀ ex *Microtus longicaudus* [AF10120]. Unuk River mouth, 56°05'N, 131°06'W, 1♂ ex *C. gapperi* [AF4359], 20-vii-93, SOM *et al.* Walker Cove, Hut Point, 55°42'48"N, 130° 54'04"W, 2♂♂, 1♀ ex *P. keeni* (2) [AF29428, 29442], 13-vii-99, AMR; same data but 1♀ ex *C. gapperi* [AF29434]. Walker Cove, Ledge Point, 55°42'20"N, 130°53'34"W, 2♀♀ ex *C. gapperi* [AF29416], 13-vii-99, AMR. Willard Inlet, inlet 2 km NW of mouth of, 54°49'N, 130° 39'W, 1♀ ex *P. keeni* [AF4299], 9-vii-93, JAC, SOM. Yakutat, 59°30'47"N, 139° 40'46"W, 1♀ ex *C. rutilis* [AF7769], 26-vii-94, CJC, AAT.

Megabothris abantis is a common vole flea in southern regions of Alaska. Holland (1958) originally grouped it with *C. charlottensis* and *C. c. protinus* because all three have a similar Pacific Coast distribution. Subsequently, he (Holland 1963: Fig. 2) classified *M. abantis* as a member of the Cordilleran Group B because it is not restricted to the coastal strip but ranges widely eastward into the Rocky Mountains (Haddow *et al.* 1983: Map 76, Holland 1985: Map 76).

Other than the closely grouped collection sites on the southeastern Alaska mainland, Holland (1958), Haas *et al.* (1980), Haas (1982), and Haas *et al.* (1982) recorded only six other localities, all northwest of the Taku River: Chilkat Peninsula, near Juneau, Klondike Highway at Moore Creek, Mosquito Lake, Taiya River, and Yakutat. Twenty-nine of our 34 new mainland records of *M. abantis* fill the large void mapped by Haas *et al.* (1989: Fig. 5) southeast of the Taku River with 16 new localities.

The recorded hosts of *M. abantis* on the mainland are *Sorex monticolus* (as *S. vagrans*), *P. keeni* (as *P. maniculatus*), *C. rutilis*, *M. oeconomus* (nests), *M. longi-*

caudus and *Z. hudsonius* (Haas 1982; Haas *et al.* 1982). Thus, three of the seven hosts of our 34 new mainland records of *M. abantis* are new for the region: *C. gapperi* (18), *M. pennsylvanicus* (1), and *S. borealis* (1). *Clethrionomys gapperi* was the most commonly collected mammal infested with this flea and produced 5♂♂ and 20♀♀ fleas, more than half of the 45 fleas (12♂♂, 33♀♀) collected. With the emphasis of the mainland survey on areas south of Juneau, only two *C. rutilis* with fleas (1♂, 1♀) were trapped. The abundance on *C. gapperi* alone, however, was concordant with the classification of host parasitism by Haddow *et al.* (1983) with the top ranking of members of the genus *Clethrionomys* along with *Microtus* as the only major hosts of this flea. Our data for *Microtus* spp., however, were insufficient for analysis with only 3♀♀ fleas ex one *M. pennsylvanicus* and one *M. longicaudus*. Consequently, the second best source of fleas from trapped hosts was *P. keeni* with 11 fleas (3♂♂, 8♀♀) from nine mice. Although *M. abantis* is not a nest flea, as indicated above with *H. o. occidentalis* and *C. charlottensis*, more specimens were found in a series of *M. oeconomus* nests collected on the Chilkat Peninsula shoreline (Haas 1982) than on a larger number of trapped hosts. Four nests were infested with *M. abantis*; of these, two had breeding populations from which were collected a total of 45 fleas (18♂♂ (9 reared), 27♀♀ (13 reared)). With the addition of our specimens, 90 specimens have been collected. The sex ratio of 1♂: 2♀♀ appears typical for *M. abantis*. Marshall (1981) calculated 30% males in a sample of 456 specimens from trapped hosts in New Mexico (Haas *et al.* 1973).

Megabothris asio megacolpus (Jordan, 1929)

Material examined: All ex meadow voles, *Microtus pennsylvanicus*. USA: AK: Klukwan, 11 km E & 12 km S, 59° 20'44"N, 135°46'11"W, 1♀ [AF8067], 30-vi-94, JAC. Klukwan, 10 km E & 9 km S, 59°21'58"N, 135°47'58"W, 1♀ [AF8081], 30-vi-94, JAC; same data but [AF8164], 2♂♂, 1♀, 1-vii-94; same data but [AF8165], 1♀; same data but [AF8185], 1♂, 2-vii-94.

Previously unknown in Alaska, *M. a. megacolpus* was first collected (three ♂♂, four ♀♀) in late June and early July 1994 by one of us (JAC) at two localities southeast of Klukwan in one of the small extensions of the range of the host, *Microtus pennsylvanicus*, from Canada into southeastern Alaska. The range of the host in this area, subspecies *M. p. alcorni* Baker, extends south from southwestern Yukon across northwestern British Columbia and into the Chilkat River valley of Alaska as far south as Haines (Miller and Kellogg 1955).

The distribution of this vole flea coincides almost completely with the range of *M. pennsylvanicus* over much of northern North America from Yukon Territory to Quebec south into the Rocky Mountains and the western Great Lakes (Haddow *et al.* 1983: Map 82; Holland 1985: Map 77). The Stikine and Taku River valleys on the southeastern Alaskan mainland also support populations of *M. pennsylvanicus* (MacDonald and Cook 1996) and may eventually yield additional specimens of *M. a. megacolpus*.

Malaraeus telchinus (Rothschild, 1905)

Material examined: Hosts *Peromyscus keeni* except as indicated. USA: AK: Gwent Cove, 54°57'00"N, 130°20'00"W, 3♂♂ [AF26560], 17-viii-98, SOM. Rudyerd Bay, 55°33'16"N, 130°51'33"W, 1♀ [AF29317], 10-vii-99, AMR; 1♂, 1♀ [AF29361], 11-vii-99, AMR; 2♀♀ [AF29379]; 2♀♀ [AF29381]; 1♀ [AF29382], 12-vii-99, AMR; same locality but 1♀ ex *C. gapperi* [AF29375], 12-vii-99, AMR; same data but [AF29378]. Rudyerd Bay, Point Louise, 55°32'42"N, 130° 53'34"W, 1♂, 2♀♀ [AF29372], 12-vii-99, AMR. Walker Cove, Ledge Point, 55° 42'20"N, 130°53'34"W, 1♀ [AF29417], 13-vii-99, AMR; 1♂, 3♀♀ [AF29458], 14-vii-99, AMR; same locality but 1♀ ex *C. gapperi* [AF29416], 13-vii-99, AMR.

The western vole and mouse flea, *M. telchinus*, eluded detection in Alaska until 3♂♂ were collected from one *P. keeni* at

Gwent Cove (across Pearse Canal from Pearse Island, British Columbia) in 1998. The next two localities were farther north at Rudyerd Bay where 2♂♂ and 9♀♀ were collected ex six *P. keeni*, and 2♀♀ ex 2 *C. gapperi*. The collector (AMR) then moved north to Ledge Point on the south shore of the mouth of Walker Cove and established the most northern locality for *M. telchinus* in North America with 1♂ and 4♀♀ ex two *P. keeni* and 1♀ ex *C. gapperi*. This new locality is ca. 236 km northwest of Kitimat, the most northern mainland locality in British Columbia (Holland 1985). Holland (1949, 1985) listed other *M. telchinus* offshore records close to southeastern Alaska on the Queen Charlotte Islands and Pitt Island. Most of his many records were clustered farther southeastward in British Columbia at inland montane and coastal localities (Holland 1985: Map 84). South of Canada these populations diverge into a coastal branch that reaches southern California and a montane branch that almost bypasses the Great Basin to reach the mesic habitats on mountains and high plateaus in Arizona and New Mexico (Haddow *et al.* 1983: Map 74).

Malaraeus telchinus is recorded from a wide range of hosts. Haddow *et al.* (1983) listed four *Peromyscus* species as the major hosts of *M. telchinus* but omitted *P. sitkensis* Merriam (= *P. keeni,* see Hogan *et al.* 1993); no *Clethrionomys* species were listed. Holland (1949, 1985) only listed four records from *C. gapperi* including one for Kitimat. The great majority of hosts on the mainland were *P. maniculatus* with *P. keeni* in the Queen Charlotte Islands. *Clethrionomys californicus* (Merriam) is a major host in Oregon; Lewis *et al.* (1988) reported more specimens of *M. telchinus* from this vole than from each of two *Peromyscus* species, three *Microtus* species, and *Lemmiscus curtatus* (Cope). This wide host range of *M. telchinus* confirms adaptability for changing major hosts when entering a region with a different fauna of potential hosts (e.g. moving from mainland British Columbia and *P. maniculatus* to mainland southeastern Alaska and *P. keeni*). We expect that *M. telchinus* occurs north of its present known northern range limit of Walker Cove because *P. keeni* occurs on the mainland north to Haines and Skagway (MacDonald and Cook 1996) and this flea occurs in "rather mesic habitats" elsewhere (Haddow *et al.* 1983: p. 108).

Opisodasys vesperalis (Jordan, 1929)

Material examined: All ex northern flying squirrels, *Glaucomys sabrinus* (Shaw). USA: AK: Chilkat River, 6.3 km WNW of Haines, 59°15'42"N, 135°33'35"W, 2♂♂, 1♀ [AF12539], 4-vii-95, CTS. Rudyerd Bay, 55°33'16"N, 130°57'33"W, 2♂♂, 3♀♀ [AF29318], 10-vii-99, AMR. St. James Bay, W side Lynn Canal, 58° 34'30"N, 135°09'30"W, 1♂, 2♀♀ [AF10306], 9-i-95, JF.

Glaucomys sabrinus, the host of *O. vesperalis*, is found along the mainland of southeastern Alaska, and on islands in the Alexander Archipelago south of Frederick Sound (MacDonald and Cook 1996). Records of its fleas are few with only one for the mainland and one for Revillagigedo Island (Haas *et al.* 1982, 1989: Fig. 7). Our three new records are the first for trapped hosts (the mainland collection near Skagway was from a nest). Lacking a pleural arch, *O. vesperalis* is a crawling nest flea (rather than a jumping flea) and remains in the nest when the host is absent (Traub 1972). Collections from other mammals are strictly accidental and rare; the flea is "essentially specific to *Glaucomys sabrinus*" (Haddow *et al.* 1983: p. 130). *Opisodasys vesperalis* is among the minority of fleas considered to be "ultraspecific", i.e., "limited to infestation of a single species of host" (Traub 1985: p. 332). It is a west coastal flea ranging from southeastern Alaska to northern California and eastward into montane squirrel habitat as far as Idaho and Montana (Haddow *et al.* 1983: Map 107; Holland 1985: Map 89; Lewis *et al.* 1988: p. 201; Haas *et al.* 1989: Fig 7).

Opisodasys keeni keeni (Baker, 1896)

Material examined: USA: AK: Chickamin River, Wolf Cabin, 55°46'N, 130°53'W, 3♂♂, 1♀ ex *Peromyscus keeni* [AF4953], 26-vii-93, SOM *et al.*; same data

but 1♀ ex *Synaptomys borealis* [AF4973]. Crescent Lake, 58°11'N, 133°19'W, 5♂♂, 7♀♀ ex *P. keeni* (4) [AF8309, 8310, 8311, 8316], 22-vii-94, SOM. Echo Cove, 58° 31'45"N, 134°54'28"W, 2♂♂, 5♀♀ ex *P. keeni* [AF21759], 21-vii-97, CTS. Gwent Cove, 54°57'00"N, 130°20'00"W, 3♂♂, 4♀♀ ex *P. keeni* [AF26560], 17-viii-98, SOM. Reflection Lake, SW side, 55° 59'59"N, 131°33'59"W, 5♂♂, 4♀♀ ex *P. keeni* [AF29116], 1-vii-99, AMR. Rudyerd Bay, 55°18'09"N, 130°50'38"W, 1♂, 4♀♀ ex *P. keeni* (2) [AF29317, 29320], 10-vii-99, AMR; same data but 2♀♀ (2) [AF29360, 29361], 11-vii-99; same data but 4♂♂, 11♀♀ (5) [AF29379, 29380, 29381, 29382, 29383], 12-vii-99; same data but 3♂♂, 1♀ (2) [AF29407, 29408], 13-vii-99. Rudyerd Bay, Point Louise, 55°32'42"N, 130°52'13"W, 2♂♂ex *P. keeni* [AF29372], 12-vii-99, AMR. Smeaton Bay mouth, 55° 18'09"N, 130°50'38"W, 1♂, 3♀♀ ex *P. keeni* (3) [AF29276, 29278, 29279], 9-vii-99, AMR; same data but 4♂♂, 1♀ [AF29292], 10-vii-99. Smeaton Bay, E Skull Creek, 55°17'27"N, 130°49'25"W, 1♂, 1♀ ex *P. keeni* [AF29283], 9-vii-99, AMR. Stikine River, Figure 8 Lake, 56° 42'N, 132°15'W, 1♂, 1♀ ex *P. keeni* [AF2627], 14-vii-92, SOM; same data but 2♀♀ [AF2628]; same data but 1♀ [AF2650], 15-vii-92, Taku River, Canyon Island, 58°33'N, 133°41'W, 1♀ex *P. keeni* [AF8272], 17-vii-94, JAC *et al.*; same data but 2♂♂, 2♀♀ [AF8273]. Unuk River mouth, 56°05'N, 131°06'W, 2♂♂, 2♀♀ ex *P. keeni* (3) [AF4355, 4360, 4428], 20-21-vii-93, SOM *et al.* Walker Cove, Hut Point, 55°42'48"N, 130°54'04"W, 3♂♂, 2♀♀ ex *P. keeni* (3) [AF29428, 29438, 29442], 13-vii-99, AMR. Walker Cove, Ledge Point, 55°42'20"N, 130°53'34"W, 1♂ ex *P. keeni* [AF29458], 14-vii-99, AMR.

This common and abundant *Peromyscus* flea is another member of the Vancouverian Group (Holland 1963) with a west coastal distribution including the Queen Charlotte Islands, extending eastward into montane habitats of its major host *P. maniculatus* in British Columbia and Alberta (Holland 1949:Map 28, 1985: Map 88). Southeast of Canada *O. k. keeni* occurs in Montana, Utah, Colorado and New Mexico (Ecke and Johnson 1952; Stark 1959; Eads and Campos 1983; Haddow *et al.* 1983: Map 108; Fagerlund *et al.* 2001; Ford *et al.* 2004). Its coastal range extends from northern California as far north as Skagway, Alaska (Haas *et al.* 1982; Haddow *et al.* 1983: Map 108; Lewis *et al.* 1988: p. 203). In British Columbia, *O. k. keeni* and *M. telchinus* have the same host, *P. maniculatus* (*P. keeni* in the Queen Charlotte Islands), and distribution (Holland 1985: Maps 84 & 88). Host sharing by these fleas also occurs in southeastern Alaska: with *M. telchinus*, *P. keeni* was host for nine collections, *C. gapperi* for three; with *O. k. keeni*, *P. keeni* was host for 37 collections (43♂♂, 55♀♀) and *S. borealis*, a new host record, was host for one (♀).

Orchopeas caedens (Jordan, 1925)

Material examined: USA: AK: 500 m S of Taiya River bridge, 59°30'11"N, 135° 20'44"W, 1♀ ex *Tamiasciurus hudsonicus* [AF12525], 1-vii-95, CTS, KDS.

The collection of 1♀ of the common, "ultraspecific" (Traub 1985) red squirrel flea, *O. caedens*, in southeastern Alaska was unexpected. The niche was already filled by *C. ciliatus protinus*, an arboreal squirrel flea well-adapted to the west coastal maritime climate on the coast and islands of British Columbia and north through the length of southeastern Alaska. Until now, there has been no record of *O. caedens* within the range of the red squirrel in this region. Elsewhere, *O. caedens* is found throughout most of the transcontinental range of the red squirrel and occurs with other red squirrel fleas, such as *Ceratophyllus vison* Baker and *Tarsopsylla octodecimdentata coloradensis* (Baker) in nests in Alaska west of the Yukon Territory (Haas and Wilson 1982). The limiting factor for *O. caedens* in southeastern Alaska is probably the high humidity and precipitation of the coastal climate. The Taiya River valley lies in the area of lowest mean annual precipitation in southeastern Alaska, i.e., less than 101.6 cm (Watson 1959).

DISCUSSION

Nineteen mammal fleas have been documented for southeastern Alaska (Haas *et al.* 1989). Three of these are known only from islands: the bat flea, *Myodopsylla gentilis* Jordan and Rothschild, and the bear flea, *Chaetopsylla tuberculaticeps* (Bezzi), on Admiralty and Chichagof Islands (Haas *et al.* 1979, 1980, 1989) and the dog flea, *Ctenocephalides canis* (Curtis), on Revillagigedo Island (Holland 1985: p. 38). Our three additions (*M. asio megacolpus*, *M. telchinus* and *O. caedens*) bring the mainland total to 19 taxa. Because of the high humidity and precipitation in the area, these new records (especially that of *O. caedens*) were not expected.

We have tried to collect the northern *Peromyscus maniculatus* flea, *Aetheca thamba* (Jordan), in the Klondike Highway pass at the Alaska/British Columbia border without success. Holland (1949: Map 40, 1958: Fig. 5, 1985: Map 73) mapped and discussed the split distribution pattern of transcontinental *A. thamba* (then *Monopsyllus thambus*). Most of the many records range from southern Yukon Territory (e.g., 1.6 km S of Carcross) eastward into northern British Columbia (e.g., Atlin) to northern and southern Alberta, northwestern Saskatchewan, and southwestern Northwest Territories. A small disjunct population exists in Quebec and Labrador. The proximity of many collections of a cold climate flea from a common and abundant host are conditions favourable for the collection of *A. thamba* in the northern Alaska/British Columbia border passes.

The bushy-tailed woodrat, *Neotoma cinerea* (Ord), is one of several wide-ranging mammals of British Columbia that has established itself in corridors of some major rivers such as the Taku, Stikine and Unuk that transect the Alaska/British Columbia border mountains (MacDonald and Cook 1996). The common woodrat flea, *Orchopeas agilis* (Rothschild) (formerly *O. sexdentatus agilis*), is the most widespread member of the *sexdentatus* group. It ranges from high mountains with the cool summers required by *N. cinerea* in New Mexico and Colorado northwestward to British Columbia (e.g., Atlin) and questionably Yukon Territory (Finley 1958; Haas *et al.* 1973; Holland 1985: Map 94; Lewis 2000; Haas *et al.* 2004). Haddow *et al.* (1983: Map 115) mapped two localities on the Alaska/British Columbia border but none in Alaska. We believe *O. agilis* probably occurs on the southeastern Alaskan mainland where *N. cinerea* occurs. Well sheltered nests should be good sources of fleas in that region.

Catallagia ioffi Scalon (formerly *C. jellisoni* Holland; see Lewis and Haas 2001) is an uncommonly collected, Holarctic mammal flea with only six known localities in Canada scattered from near Dawson in Yukon Territory southeastward through British Columbia (e.g., Atlin) to Banff National Park, Alberta (Holland 1954; Hopkins and Rothschild 1962; Haas and Johnson 1981; Holland 1985 :Map 17). The likelihood of collecting specimens on the southeastern Alaskan mainland is suggested by this elongated distribution pattern in western Canada as well as the diversity of known hosts, e.g., *P. maniculatus*, *N. cinerea*, *C. rutilis*, *C. gapperi*, *M. pennsylvanicus* and *Lemmus trimucronatus* (Richardson). The latter species has not been recorded for southeastern Alaska (MacDonald and Cook 1996).

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SCIENTIFIC NOTE

Infestation of Bent Grass by a New Seed Pest, *Chirothrips manicatus* (Thysanoptera: Thripidae), in Oregon

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Bent grasses (*Agrostis* spp.) (Poaceae: Pooideae) are used extensively on golf courses. Oregon is the largest bent grass seed producing state in the U.S.A., with over 3,900 ha under cultivation of *Agrostis stolonifera* L. (= *Agrostis palustris* Hudson) (creeping bent grass), *Agrostis castellana* Boissier & Reuter (dryland or Highland bent grass) and *Agrostis capillaris* L. (= *Agrostis tenuis* Sibthorp) (colonial bent grass) (USDA-ODA 2002).

In June 2004 *Chirothrips manicatus* Haliday (Thysanoptera: Thripidae) were detected within individual florets of *Agrostis* spp. in Oregon. Voucher specimens were deposited in the Oregon State Entomology Museum, Corvallis, OR. Each floret with a thrips produced no seed.

Chirothrips manicatus is widespread in North America. In Oregon it has been collected from flowers of various plants (Post 1947), but this is the first report of it developing in florets of *Agrostis*. It has been reported on *A. tenuis* in New Zealand (Mound and Walker 1982) and on *Agrostis* sp. in Europe (zur Strassen 2003), but there is no information on its impact on seed production on these hosts. It is reported as a pest of orchard grass in New Zealand (Doull 1956).

To determine the extent to which *C. manicatus* was present in commercial *Agrostis* seed production fields in Oregon, we surveyed 13 bent grass seed production fields in July 2004 (Table 1). The fields were located in the Silverton Hills area in

Table 1.

Incidence of *Chirothrips manicatus* in *Agrostis* seed production fields in the Willamette Valley in western Oregon. Means ± SE are based on collections of 17 to 50 panicles from each of four transects in a diamond pattern in each field.

Field	*Agrostis* host	Cultivar	Mean % panicles with thrips	Mean no. thrips per panicle	Mean no. seeds per panicle	Mean % seed loss due to thrips
1	*A. castellana*	Highland	87.9 ± 4.1	22.6 ± 2.1	452 ± 105	5.1 ± 1.4
2	*A. castellana*	Highland	49.8 ± 7.3	6.8 ± 0.9	426 ± 53	0.9 ± 0.2
3	*A. castellana*	Highland	60.0 ± 5.7	10.3 ± 1.5	526 ± 55	1.1 ± 0.4
4	*A. castellana*	Highland	26.2 ± 5.4	5.4 ± 2.5	233 ± 20	1.3 ± 0.7
5	*A. castellana*	Highland	80.5 ± 7.1	17.9 ± 2.3	375 ± 11	4.1 ± 1.8
6	*A. castellana*	Highland	32.9 ± 4.3	12.5 ± 3.2	471 ± 10	1.0 ± 0.4
7	*A. castellana*	Highland	64.8 ± 10.1	17.6 ± 2.0	467 ± 9	2.8 ± 0.5
8	*A. castellana*	Highland	38.0 ± 7.8	6.0 ± 0.7	277 ± 22	0.8 ± 0.3
9	*A. stolonifera*	Crenshaw	2.0 ± 1.4	4.8 ± 2.5	528 ± 44	0.02 ± 0.01
10	*A. stolonifera*	Princeville	9.5 ± 3.1	1.4 ± 0.1	452 ± 22	0.03 ± 0.01
11	*A. stolonifera*	Pennlinks	0	0	321 ± 11	0
12	*A. stolonifera*	Penncross	0	0	292 ± 12	0
13	*A. capillaris*	Alistar	0.5 ± 0.5	0.03[1]	425 ± 13	0.01 ± 0.01

[1] Five individuals in a single panicle

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the Willamette Valley in western Oregon. In each field, 17 to 50 panicles were collected at random along each of four transects in a diamond pattern. Panicles were examined under a stereo microscope and the number of thrips recorded (Table 1). Seeds from each panicle were threshed by hand to avoid seed loss. Caryopses were separated from the lemma and palea using a scarifier (Model PSS1000, Mater International, Inc., Corvallis, OR) and debris was removed with an air column (Alderman *et al.* 2003). Total seed weight from each transect was determined and the weight of a subset of 200 seeds from each transect was used to estimate the total number of healthy seeds in panicles collected from each transect. Percentage seed loss was estimated as: [number of infested seeds / (number of infested seeds + estimated number of healthy seeds)] x 100, where the number of infested seeds equals the number of thrips, based on our observation of one thrips per floret and destruction of a single seed by each thrips.

Overall, 32.5% of 2,310 panicles from the 13 fields that were examined were infested with thrips and the abundance of the thrips appeared to be linked to the host (Table 1). The greatest infestation was observed in Highland bent grass (*A. castellana*) which is the most common cultivar grown for seed in Oregon. Individual bent grass florets contained a single *C. manicatus* pupa or adult (apterous male or winged female) with its head towards the base of the floret (Figure 1). The thrips were enclosed firmly between the lemma and the palea, and were not easily dislodged. In florets where a thrips was present, organic debris was visible but there was no trace of the caryopsis (seed). These data negate previous speculation that small seeded plants such as *Agrostis* spp. are unlikely hosts for *C. manicatus* (Doull 1956).

It is not known how long *C. manicatus* has been present on *Agrostis* in Oregon. As there is no external indication of *C. manicatus*, it is possible that its presence could have been undetected. Female *C. manicatus* overwinter within florets in the field (Doull 1956) and therefore it is also possible that *C. manicatus* has emerged as a pest due to the phase-out of field burning in the late 1980's.

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Figure 1. *Chirothrips manicatus* within a floret of *Agrostis castellana*.

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SCIENTIFIC NOTE

Comparative Activity of the Codling Moth Granulovirus Against *Grapholita molesta* and *Cydia pomonella* (Lepidoptera: Tortricidae)

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ABSTRACT— The granulovirus of codling moth, *Cydia pomonella* L., CpGV, is now commercialized for codling moth control in pome fruit in the USA and Canada. It is highly specific for codling moth and related species. Comparative assays of CpGV against neonate larvae of another introduced tortricid pest, the oriental fruit moth, *Grapholita molesta* Busck, revealed a 557 and 589 fold lower susceptibility of neonate larvae compared with the LC_{50} and LC_{95} values derived for *C. pomonella*.

Since its introduction into North America, the oriental fruit moth, *Grapholita molesta* Busck, has become a widely established pest of peach, nectarine, apricot, and apple (Rothschild and Vickers 1991). There is little information regarding naturally occurring disease of the oriental fruit moth, with the exception of microsporidia in adults (Simchuk and Komarova 1983) and *Bacillus thuringiensis* in larvae (Grassi and Deseö 1984). Although field trials with various formulations of *B. thuringiensis* have been reported for oriental fruit moth, results indicate that it is relatively ineffective (Rothschild and Vickers 1991).

Following its initial discovery in infected codling moth *Cydia pomonella* L. larvae in Mexico in 1964, numerous laboratory and field studies have confirmed the virulence of the codling moth granulovirus (CpGV), against its homologous host (Falcon *et al.* 1968, Laing and Jaques 1980, Arthurs and Lacey 2004, Cossentine and Jensen 2004). In early host specificity studies, CpGV was also noted to have larvicidal activity against the pea moth, *Cydia nigricana* (Fabricius) (Payne, 1981) and oriental fruit moth (Falcon *et al.* 1968), but quantitative assays of the virus have not been reported for the latter species. We conducted bioassays of the Cyd-X formulation of CpGV (Certis USA, Columbia, MD) against oriental fruit moth and codling moth neonates from colonies maintained at the Yakima Agricultural Research Laboratory using the materials and methods described by Lacey *et al.* (2002). The codling moth diet described by Brinton *et al.* (1969) (BioServ, Frenchtown, NJ, USA) was used for both species.

Following initial bioassays to determine mortality ranges, five concentrations of Cyd-X that produced mortality in neonate larvae ranging from 10 to 96.7% in oriental fruit moth and 36.7 to 96.7% in codling moth were bioassayed against 30 neonates per concentration. Bioassays were conducted on artificial diet in 2-ml plastic conical autosampler vials (Daigger, Lincolnshire, IL, USA). A 2-mm diameter hole in the cap of each vial covered with stainless steel screen (0.16 mm mesh size) eliminated condensation. Ten µl of aqueous virus suspensions or 10 µl of water for controls was applied to the surface of 1 ml of artificial medium (approximately 100 mm^2) in the autosampler vials. The label specified virus concentration of Cyd-X is 3 x 10^{13} granules per liter. After the surface of the medium had dried, one neonate larva was added to each vial. The vials were incubated for 7 d at 25 ± 1.7 °C and then assessed for larval mortality. The study was repeated for each species on four separate dates. Each date was treated as an individual replicate of each concentration (i.e. data were not pooled before probit analysis).

The results of the assays clearly indicated that oriental fruit moth neonates are susceptible to CpGV, but at a significantly lower level than that observed in codling moth neonates (Table 1). The oriental fruit moth were 557 and 589 times less susceptible to CpGV compared with codling moth, based on probit (normal sigmoid) analysis of the LC_{50} and LC_{95}, respec-

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tively (StatsDirect Ltd, v. 2.4). Based on our methods, the calculated LC_{50} and LC_{95} of CpGV for oriental fruit moth are 35 and 540 granules per mm^2, respectively, but only 0.06 and 0.9 granules per mm^2 for codling moth. Although CpGV must be ingested in order to infect a larva, Ballard *et al.* (2000) demonstrated that the point of entry of codling moth larvae into fruit may not necessarily be where virus is acquired; larvae could become infected by walking or browsing on CpGV-sprayed leaf surfaces in as little as 3.5 min. Ostensibly virus picked up on legs or mouth parts in the absence of browsing leaf surfaces could contaminate the initial point of entry. In our bioassays, neonates of both species may wander over the surface of the medium before boring into it. In the case of codling moth larvae this would provide ample opportunity to acquire virus even at the lower concentrations. Huber (1986) estimated that the LD_{50} for neonate larvae could be as low as 1.2 granules per larva.

First generation oriental fruit moth often feed on shoots and young foliage. Although not as active against oriental fruit moth as against codling moth in laboratory bioassays, field activity of Cyd-X against oriental fruit moth neonates at label rates used for codling moth control (0.07-0.44 L/ha) could potentially reduce oriental fruit moth populations if significant feeding of early instars of the first generation occurred on treated foliage. Because natural feeding behavior will influence their susceptibility to CpGV, further field studies are warranted.

We are grateful to Rob Fritts Jr. (Certis) for Cyd-X samples and the Washington Tree Fruit Research Commission for financial support. We thank Don Hostetter and Joel Siegel for helpful reviews of the manuscript.

Table 1.

LC_{50} and LC_{95} values for CpGV bioassayed against *Grapholita molesta* and *Cydia pomonella* neonates. The number of granules per 10 µl is based on dilutions of Cyd-X with a label specified virus concentration of 3×10^{13} granules/L. All probit comparisons were significantly different, $P < 0.001$ based on dosage $\log_{(10+1)}$ and adjusted for control mortality (< 7.1%).

Species	n	LC_{50} (95% CI)	LC_{95} (95% CI)	Slope
C. pomonella	714	6.30 (4.74 – 7.91)	91.62 (63.00 – 155.68)	1.41
G. molesta	708	3.51×10^3 ($2.72 - 4.42 \times 10^3$)	5.40×10^4 ($3.66 - 9.04 \times 10^4$)	1.39

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SCIENTIFIC NOTE

A novel host association for *Monarthrum scutellare* (Coleoptera: Curculionidae: Scolytinae) in British Columbia

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Monarthrum scutellare (LeConte) is an ambrosia beetle that ranges from British Columbia to northern Baja California in Mexico (Bright and Stark 1973, Wood 1982, Wood and Bright 1992). It is recorded to breed in various species of Fagaceae including *Chrysolepis*, *Lithocarpus densiflora* (Hooker & Arnold) Rehder, *Quercus* spp., *Quercus agrifolia* Neé, *Q. garryana* Douglas and *Q. kellogi* Newberry (Farris 1965, Bright and Stark 1973, Bright 1976, Wood and Bright 1992) with single records from *Abies* and sequoia that Bright and Stark (1973) considered accidental or erroneous.

On 2 May 2005, a 38-cm length of "green" split alder firewood and associated Scolytinae (Coleoptera: Curculionidae) collected from a recently delivered commercial load of firewood were submitted to the Canadian Forest Service for identification after beetles were observed emerging from the wood. The half stem section was split off-centre, included all annual growth rings, and was 18.2 cm in diameter and 27 years of age. No bark was present on the piece of firewood, however, a single V-shaped parental gallery 22 mm in length was incised in the sapwood and five adult *Alniphagus aspericollis* (LeConte) (Curculionidae: Scolytinae) were associated with the sample. The gallery shape agrees with those described by Bright and Stark (1973) as typical for *A. aspericollis*. The presence of a parental gallery of *A. aspericollis* and the structure of the wood (absence of rays, ring porous wood) confirmed that the host attacked was *Alnus rubra* Bongard (Betulaceae).

Boring dust was being actively extruded from ambrosia beetle galleries along the split face of the wood; however, no entrance holes were observed on the outer face of the bole. The wood was held at room temperature for adult emergence and 52 female and 55 male *M. scutellare* emerged between 2 May and 17 May 2005. The sample was then split longitudinally and the distribution of galleries along the split face enumerated by growth ring and growth ring widths measured to the nearest 0.5 mm. All of the 22 *M. scutellare* galleries visible on the split face were in the widest growth rings (mean ± SD = 5.6 ± 0.99 mm) from the first nine years of growth. No galleries were apparent in the outermost 40.5 mm of the xylem comprising the last 18 years of growth.

A band saw was used to cut 1-2 cm thick cross-sections containing ambrosia beetle galleries and the galleries dissected. Bifurcations were evident in four of the five partial galleries dissected, with three having a single bifurcation and one bifurcating twice. The galleries dissected (longest arm) ranged from 12.5 to 52.8 mm in length and were heavily stained black, likely by the ambrosia fungus introduced by the female beetles (Farris 1965). Although larvae of *Monarthrum* species, including *M. scutellare* (Wood and Stark 1973), *M. mali* (Fitch) and *M. fasciatum* (Say) (Solomon 1995) characteristically develop in "cradles" excavated above and below the sidewalls of the parental galleries, no brood cradles were evident in the dissected galleries or on the radial faces of the split wood. While no evidence of brood production was found during gallery dissections, the heavy staining observed along the length of the dissected galleries indicates that the observed attack was not recent and the large

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numbers of adults recovered suggests that *M. scutellare* can attack and breed in red alder. At least one oak-feeding species of *Monarthrum*, *M. laterale* (Eichhoff), has been recorded from alder and *M. bidentatum* Wood, *M. hoegi* (Blandford) and *M. umbrinum* (Blandford) breed in *Alnus* spp. (Wood 1982, Wood and Bright 1993). Thus, while *M. scutellare* is usually associated with species of Fagaceae, it is possible that hosts in other families may also be utilized. Alternatively, the emergent adults could represent mature adults attempting to establish brood in the firewood piece. Because galleries associated with this collection were incomplete, the ability of *M. scutellare* to develop in *A. rubra* cannot be determined with certainty. The presence of brood production in choice and no-choice breeding trials of *M. scutellare* in cut stem sections of native Fagaceae (*Quercus garryana* Douglas ex Hooker) and *A. rubra* or the discovery of brood in naturally attacked red alder will be necessary to confirm breeding in non-traditional hosts. Although evidence of breeding of *M. scutellare* in red alder is currently circumstantial, such novel host associations have been demonstrated to occur in other ambrosia beetles. Nijholt (1981) reported attack in red alder by two species of ambrosia beetles, *Gnathotrichus retusus* LeConte and *Trypodendron lineatum* (Olivier), which normally utilize coniferous species as hosts (Bright 1976, Wood and Bright 1992). Kunholz *et al.* (2000) subsequently confirmed red alder as a breeding host for *G. retusus*, while Lindgren (1986) documented brood production by *T. lineatum* in bigleaf maple, *Acer macrophyllum* Pursh.

Voucher specimens of *A. aspericollis* and *M. scutellare* have been deposited in the reference collection at Canadian Forest Service, Pacific Forestry Centre, Victoria, British Columbia. L. Safranyik, T. Shore and A. Carroll, Natural Resources Canada, Canadian Forest Service reviewed an earlier version of this manuscript. Their helpful comments and those of two anonymous reviewers are gratefully acknowledged.

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SCIENTIFIC NOTE

Notes on the status of the Eurasian moths *Noctua pronuba* and *Noctua comes* (Lepidoptera: Noctuidae) on Vancouver Island, British Columbia

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Two Eurasian cutworm moths (Lepidoptera: Noctuidae), *Noctua pronuba* (Linnaeus) and *Noctua comes* (Hübner), both accidentally introduced to North America, are now sympatric in southwestern British Columbia. The former was first recorded in Nova Scotia, the latter in British Columbia. This paper reports the occurrence of both species for the first time on Vancouver Island. They are the only species of the genus *Noctua* known in North America (Lafontaine 1998).

Noctua pronuba (the large yellow underwing) was first reported in North America in Nova Scotia in 1979 (Neil 1981). It is now known from every Canadian province and Nunavut (Troubridge and Lafontaine 2005) and, in the USA, from Maine (Wright 1987) to Louisiana (Brou 1997) and California (Powell 2002). It was first recorded in BC in 2002 (CNC [Canadian National Collection of Insects and Arachnids, Ottawa] data) and is now abundant on eastern Vancouver Island as far north as Sayward (RBCM [Royal British Columbia Museum, Victoria] data) and areas of the lower Fraser River Valley (K. Needham, pers. comm.). We have yet to hear of any records from the BC Interior.

Noctua comes (the lesser yellow underwing) was first recorded in Canada in Burnaby, BC in August 1982 (Neil 1984) although a specimen in the Spencer Entomological Museum, UBC (University of British Columbia, Vancouver) was collected in Vancouver in July 1982. This species was first recorded on Vancouver Island in Victoria in 1990 (PFC [Pacific Forestry Centre, Victoria] data) and is now abundant in suburban habitats there. Elsewhere in BC *N. comes* has been found in the Okanagan Valley and Lillooet and south to Oregon (Lafontaine 1998, J. Troubridge, pers. comm.).

Noctua pronuba has a wingspan of 50-60 mm and a diagnostic orange-yellow hindwing with a broad black border. Images of adults and larvae are in Wright (1987), Lafontaine (1998), and Neil and Specht (1987). *Noctua comes* is similar but normally has a black mark near the centre of the orange of the hindwing; genitalia differences distinguish the two species unequivocally (Lafontaine 1998).

Although *N. pronuba* is known to be migratory and a very strong flier (Passoa and Hollingsworth 1996), its spread may have been facilitated by human activity. It has a wide range of host plants, many of which are part of the horticultural trade, food-crop industry, or are widespread weeds. Host plant genera include: *Holcus* (J. Tatum, pers. comm.), *Poa* and other grasses (Wright and Neil, 1983), *Atriplex, Chrysanthemum, Dianthus, Fragaria, Freesia, Gladiolus, Myosotis, Polygonum, Primula, Ribes, Stellaria, Taraxacum* and *Viola.* Larvae also eat various common food crops (Passoa and Hollingsworth 1996, B. Duncan, pers. comm). *Noctua comes* has been recorded on *Conium, Cornus, Potentilla, Calendula, Cardamine, Cirsium, Digitalis, Fragaria, Myosotis, Plantago, Primula* but, most often, on *Rumex crispus* (J. Tatum, pers. comm.) as well as tobacco and grapes (Sannino and Espinosa 1999) and *Crataegus* (Ward 2003).

Life-history details of *N. pronuba* are reported in Singh and Kevan (1965),

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Wright and Neil (1983), Morris (1987), and Passoa and Hollingsworth (1996). In British Columbia, *N. pronuba* will likely exhibit the univoltine life history typical of European and eastern North American populations.

Each female lays up to 2000 eggs on leaf undersides (Morris 1987) or non-host substrates (B. Duncan, pers. comm.). Larvae feed on foliage, crowns and roots of hosts; immature larvae usually overwinter (Morris 1987), but in coastal BC mature larvae also do so (B. Duncan, pers. comm.). Mature larvae pupate in the soil in the spring (Passoa and Hollingsworth 1996). Tachinid flies parasitize *N. pronuba* in eastern North America (J. Troubridge, pers. comm.), but have not been recorded in BC on *N. comes* or *N. pronuba* (J. Tatum, pers. comm.). However, *Trichogramma* wasps parasitize egg masses in the province (B. Duncan, pers. comm.).

As it is a strong, migratory flier (Passoa and Hollingsworth 1996), can endure very cold winters (Wright and Neil 1983), and feeds on a wide array of plants associated with humans, *N. pronuba* will probably colonize all of BC. Adults may lay eggs on non-plant substrates (B. Duncan, pers. comm) or hide during the day in objects around human habitation (although they fly readily when disturbed), making them good candidates for transport by vehicles.

Because of its growing abundance in coastal BC, we believe that *N. pronuba* may become an economic pest, although in the long-term, populations will likely be moderated by increasing parasitism. *Noctua comes* will probably have a similar future.

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